

**HERPETOLOGICAL COLLECTING
AND COLLECTIONS MANAGEMENT**



JOHN E. SIMMONS

SOCIETY FOR THE STUDY OF AMPHIBIANS AND REPTILES

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NO. 16**

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BY

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INTRODUCTION

This handbook is for anyone who needs to collect and preserve amphibians and reptiles or who works with preserved specimens in a collection of any size.

Each systematic herpetological collection has its own peculiarities, strengths, and weaknesses. There are no established, standard procedures for managing a collection, but since the pooled resources of many collections are often needed for research, there is a need for common standards for curatorial practices. This handbook provides a framework for standardizing curatorial practices among diverse collections. The framework is flexible enough to accommodate everything from a large, major museum collection to a small teaching collection.

This handbook is also intended to give a complete understanding of what happens to a specimen from capture through cataloging. I believe that if more field workers understood what happens to the specimens in the museum, they could make more useful collections of specimens. If more users of preserved collections understood how the collections are managed, there would be fewer problems when material is borrowed.

Perhaps most importantly, this handbook is for those who are suddenly faced with the need to learn field collecting techniques or collections management techniques who have not had the opportunity to be exposed to them before.

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The participants in the 1987 Collections Care Pilot Training Program at the Los Angeles County Natural History Museum contributed ideas, criticisms and encouragement but most of all, made me aware of just how little we really know about conservation of natural history collections.

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Lastly, my thanks go to the curator who gave me my first job working in a museum, my first chance to do real field work, and who has continued to keep my career in herpetological collections management filled with new opportunities. In appreciation for his support, this work is dedicated to William E. Duellman.

FIELD COLLECTING

"One of the most important aspects of frogs and salamanders is their martyrdom to science."

—G.K. Noble (1931)

Introduction

Not too many years ago, in the heyday of the growth of natural history museums, professional collectors were employed to bring back all manner of beasts to the distinguished scientists who waited in their laboratories. As collection growth has slowed, many of the skills necessary for efficient field work have been lost (along with the full-time collectors). At the same time, the standards for field collections have become more sophisticated to meet the demands of systematic biology. The result is that more is expected of a field collector than ever before, yet there are fewer opportunities to learn good field methods. With that in mind, this section will focus on some techniques and general recommendations for collecting, preserving, and documenting reptiles and amphibians in the field.

Documentation of the collection

Good record keeping starts with good field notes. The importance of taking good field notes cannot be exaggerated (Smith 1975). Notes should be made on 100% cotton rag paper with an acceptable ink (see Williams and Hawks 1986 for a comparison of inks). Do not use pencil or ball-point pen, as these both fade more than permanent inks. Each entry should include the following information:

1. Locality, given in standard format (Riemer 1954).
2. Date and time of collection (date of preservation if different).
3. Field number (a field tag must be attached to each individual specimen) and name of collector(s).
4. Description of specimen, especially colors and other features likely to be lost or destroyed by preservation (Smith 1955). The use of a standard color guide, such as Smith (1975) is highly recommended since colors are notoriously difficult to describe (McDiarmid, 1982).
5. Habitat the specimen was found in.
6. Other material related to the individual specimen, such as photographs and tape recordings.

The format for field notes may vary according to the requirements of a particular situation (see Lambiris 1979 and Rubio 1968 for examples). Three-ring loose-leaf binders are the most useful to carry in the field. Bound books restrict the number of pages available and prevent the insertion of additional notes and maps. It is also very unwise to take original copies of previous field notes into the field. Loose sheets are too easily lost. Tape recording of field notes has many drawbacks, ranging from accidental erasure to the limitations of the spoken word for descriptive purposes. Tape recordings may be used to amplify field notes, but should never be used as a substitute for them.

Field tags affixed to specimens should be relatively small and of sturdy stock. Everything has been used, from cheap paper to wood and plastic, with varying degrees of success. Metal tags should be avoided since many kinds of metal will corrode in preservatives, and metal tags are usually heavy enough and sharp enough

to damage specimens. Restrict the content of the tag to the initials of the collector and a unique number for each individual specimen. Attempts to devise a numbering system which incorporates the year and date of collection of the specimens result only in confusion for others trying to use your field notes (long after you are dead and preserved yourself), and compound the chance of a copying error occurring during data management processes. When a continuous sequential numbering system is employed, year and date of collection will be obvious to anyone familiar with the field series anyway.

The use of tags with the complete locality on them is redundant when proper notes are kept. This kind of tag is also clumsy to work with and likely to damage specimens and then disintegrate long before a smaller, sturdier tag.

Permits

Planning a collecting trip begins with acquiring the appropriate permits to collect, export, and import specimens. Most states now require permits for scientific collection of amphibians and reptiles (Ashton 1976; Czajka and Nickerson 1974; Dodd 1976), and you will **definitely** have to produce documentation to bring a collection in to the United States from a foreign country. Refer to Berger and Phillips (1977) and Berger (1980) for assistance in determining just what you will need.

MAKING THE COLLECTION

“Bad work in collecting is, in nine cases out of ten, due to one of two causes—ignorance or laziness. By some curious process of reasoning, many really intelligent men conclude that they can go into the field and collect successfully without having learned a single thing about methods, or asked a word of advice from a competent instructor.”

W.T. Hornaday (1905)

Time to collect

When to collect is often dictated by habitat type or scheduling restrictions with regard to both day vs. night and wet vs. dry seasons, but collecting at the “wrong” times can often yield animals otherwise overlooked. Don’t let preconceptions about the area to be worked limit the choice of equipment and field clothing before departure for the trip. Go prepared to work day and night, rain and shine.

In general, for seasonal conditions, wet-season collecting is more profitable than dry-season, although in many areas the dry season has the effect of concentrating the wildlife about dwindling water resources, thus increasing chances of obtaining some species.

How Much to Collect

Once in the field, a balance must be achieved between the number of specimens collected and the time available for their preparation. If more than a couple of collectors are working in an area, it is very easy for the tide of incoming specimens to overwhelm the cataloging and preserving process (you can tell this has happened when the expedition leader, eyes studded red by formaldehyde fumes, staggers from the processing area and confiscates all the plastic bags in camp). As a general rule,

it will take approximately 5-10 minutes per specimen for note taking, killing, and preserving, as an absolute minimum (usually longer).

Be sure to collect eggs, larvae, and juveniles as well as adults, as this material is not found in most museum collections (Alberch 1985).

When preparing for a collecting trip, take the time to carefully look over any material from the area you will be going to which is already in museums. There is no excuse for collecting animals which are not needed.

Places to Collect

Most collections being made in the field at present are associated with some particular project, such as voucher specimens for an ecological study, or specimens selected to monitor reproductive activity of organisms in a community. For this type of collecting, the locality is dictated by the requirements of the study.

For general survey work, when a sampling of all species in the area is desired, collecting should be approached with an open mind. By day, turn over rocks, logs, and pieces of metal or wood lying on the ground. By night, walk likely trails with a light, and check ponds, swamps, and streams. Many animals which are difficult to collect during the day, like lizards, can be plucked easily from their perches at night (Hoffmeister 1951). Most collectors are familiar with the "edge effect" phenomenon—animals often are more plentiful at the edge of disturbed situations than they are in virgin habitat. Hunting along roads, fence rows, around habitations, trash dumps, wells, irrigation ditches and the like is often quite productive. In urban areas, check parks, trash piles, and even inside sunken utility meter boxes (McCoy 1961).

Following are some typical situations and suggestions for working them:

ROAD CRUISING. This is the classic technique for the snake hunter in the Age of Automobiles (Campbell 1956; Klauber 1935; Ruick 1948). If seldom traveled roads are available, especially blacktop, cruising at slow speeds from dusk on is a good way to find specimens which crawl out on the road for warmth. Be prepared to leap from the vehicle quickly, and take a flashlight with you when you do. A spotlight may be used to search for creatures on the sides of road cuts. There are a variety of powerful lighting devices available which plug into the cigarette lighter of the car. Be warned, however, that their use is illegal in some place and road cruising itself is illegal in one state.

STREAMS. Night collect by walking along the bank or in the stream with a headlamp. Check exposed rocks in the stream and the overhanging vegetation as well as stream banks. For day work in streams, rolling rocks over in the stream bed is often successful, as is dip-net work, especially in areas where stream flow is diminished. Many amphibians hide in cavities created by streams which undercut their banks. Amphibian larvae blend in well with stream bottoms and hide among the gravel and detritus. Some, equipped with sucker mouth parts, clasp to the undersides of rocks, and can be caught by a downstream net when rocks are moved (Svihla 1959).

HILLSIDES. Flip rocks. This is usually more successful on slopes of 45 degrees or less. Thinner, flatter rocks generally are preferred over thicker ones. Be alert for eggs as well as moving beasts beneath the rocks. Always return flipped rocks and rolled logs, etc., to a close semblance of their original condition.

PONDS. Pond edges should be worked with a light by night. It is a wise idea to check out the pond in daylight first, however. By day, the use of a dip net, seine, or turtle traps is recommended. Check also man-made structures at ponds and lakes (Hawken 1951). Large frogs may even be taken on lures or flies (Miller 1972).

DESERTS. Road cruise (see above) and look around rocky areas with a light by night for lizards. By day, flip rocks and debris, and search cracks in exfoliating boulders (Morrison and Tanner 1966) and between rocks. Spring rains in the desert (when creatures run riot in temporary pools) are the most productive times to collect, and may be the only time you will find most amphibians. You might also try trapping (Banta 1957). On fine-textured soils, snake tracks (Lillywhite 1982) can be followed.

STREETLIGHTS. Being a magnet for insects, streetlights are also a beacon for other wildlife such as toads.

ROADSIDE DITCHES AND TEMPORARY POOLS. These ephemeral habitats often harbor exotic things like frogs and the snakes which come to eat them, especially immediately after a rain,.

SWAMPS. Best collected for most species at night with a light source, or seined, netted or trapped for larvae and turtles.

TROPICAL FORESTS. By day, these are extremely difficult to work, except for sifting leaf litter and log rolling. Myers (1956a) describes a method of catching small lizards on the forest floor by dangling an insect tied to a thin line on the end of a pole in front of likely looking places among leaf litter and debris. The best collecting technique is walking trails at night with a light source, preferably a headlamp (which will catch eye-shine better than hand-held sources). Steep-sided trenches (about 0.5-1.0 m deep) can be dug to trap specimens in areas where the collector will be in residence for a lengthy period of time. Be sure to cave in the sides of any trenches when collecting in an area is completed so they do not continue to trap animals after you are gone. Digging in damp areas and seeps is said to be a good way to turn up caecilians. Edward H. Taylor, when asked his secret for using this method so successfully, offered me this advice: "Don't waste your time digging where you aren't going to find anything." The tropical forest vegetation can be quite overwhelming at first, and it takes some time to form a "search image" of your prey in your mind. Remember also to look from the forest floor up into the trees as you move along slowly and not develop the habit of just looking at eye level. It is amazing how many fieldbooks are filled with records of specimens "found 1.5 m above forest floor" and few below or above.

LEAF LITTER. A small rake or machete blade can be used to sift systematically through leaf litter on the forest floor by day. Eggs of both reptiles and amphibians are turned up this way as well as adults.

CAVITIES. In the tropics, holes in trees, stream banks, and the water collected in agaves, bromeliads and other plants are all excellent places to search for small reptiles and amphibians. Eggs are sometimes found in cavities in bamboo, hollow trees, and epiphytic plants.

TREE FALLS. Both natural and man-induced, tree falls often yield seldom collected species both in the roots and among the upper branches.

TREE TOPS. For the adventurous or well-insured, the canopy of the forest can be accessed using modified rock-climbing techniques and lots of rope. Refer to Bowles (1911), Hingston (1932), North (1943) and Perry (1980) for the thrilling details. A technique has even been developed for using drift fences in trees (Vogt 1987).

AREAS BEING BURNED OR CLEARED. Many forms of wildlife flee encroaching fires, and many more are to be found by following bulldozers about at construction sites. This technique yielded good results for Duellman (1978) and his crew at Lago Agrio in eastern Ecuador.

USE OF LOCALS. If you will be in one place for a reasonable length of time, it is often profitable to offer monetary remuneration for specimens. Resident children are usually far better local collectors than the visiting field person. Herpetologists have even gone so far as to broadcast their desires for specimens over the local radio station (Shaw 1962). Careful questioning will be necessary to obtain the proper field documentation concerning the location of the individual specimen when collected by someone else. Price adjustments must usually be made over time to discourage over-collecting of common species. Other local life forms may also aid in the location of desirable specimens, as Smith (1946) discovered with flocks of West Texas turkeys.

DRIFT FENCES. Along with can traps, pit traps, and trenches, these can be used when an extended stay is expected in an area (Figure 1). They can be made of metal, cloth, plastic, or wood. One novel trapping technique used on lizards was to toss a hat on the ground and allow the beasts to creep under it (Klein 1951). For more details on trapping techniques, refer to Boarder 1969; Bourke 1968; Campbell and Christman 1982; Carpenter 1953; Clark 1966; Cockburn et al. 1979; Dargan and Stickel 1949; Fitch 1951; Franklin 1947; Hall 1970; Heatwole et al. 1965; Jackley 1947; Lannon 1962; Middendorf 1979; Moulton 1954; Parker 1971; Rodgers 1939; Smith 1971; Vogt and Hine 1982; Vogt 1941; Vogt 1987; and Whitaker 1967a.

EGGS. Eggs of both reptiles and amphibians should be hatched if it is possible under the field conditions to which you find yourself subjected. Reptile eggs can be incubated in their native soil in a closed plastic bag opened occasionally for air, or by the method detailed by Tryon (1975) if materials are available. Amphibian eggs can also be incubated in plastic bags, but be sure to change the water often if it is not aerated. Amplectant pairs of amphibians may be kept in plastic bags until eggs are deposited, then the eggs hatched. If you are in the fortunate position of being able to raise larvae through metamorphosis, pickling a few at various stages (see Gosner 1960; or Duellman and Trueb 1985 for developmental stages of larvae) will yield a very useful developmental series. For many tropical anurans, this type of collection can fill in important gaps between unknown tadpoles and known adult frogs. Feed developing larvae on stream or pond detritus from the water source where the adults or larvae were collected.

OCEANS. Sea turtles are notoriously hard to collect except when ashore to deposit eggs, but you can always go after them with a line tied to a remora (De Sola 1932).

Further ideas for collecting a variety of creatures may be found in the following references: Bhargava 1970; Bishop and Schoonmacher 1921; Brown 1971; Chaney and Smith 1950; Conant 1951, 1975; Cooper and Poulson 1968; Jorgensen and Orton 1961; La Rivers 1974; Marrone 1978; Mount and Schwaner 1970; Murphy 1968; Rathor 1970; Schmidt and Davis 1941; Schoenhoff 1974; Smith 1978; Stebbins

1985; Valentine 1963; Visser 1971; Webb and Messel 1977; Whitaker 1967b; Whitlock 1971; and Wilson 1971.

Equipment

When traveling out of the country to collect, never plan on purchasing supplies locally unless you know you will be passing through a large city and have plenty of time on your hands. The simplest items can be the hardest to locate in a strange place (describing a dip net in Spanish can be an exasperating experience).

Following is a list of some suggested equipment for field work:

CLOTH AND PLASTIC BAGS OF VARIOUS SIZES. It would seem that you can never have enough of either of these. Make sure cloth bags have double-sewn seams and are deep enough to be tied at the top once the snake or lizard is safely inside (see also Huheey 1964). Plastic bags must have air-tight seams.

LONG FORCEPS. The foot-long variety, or placental forceps with a scissor grip are very useful with a multitude of field applications both for collecting and preserving specimens.

NETS. Includes dip nets (for tadpoles, straining out larvae and eggs from samples of gunk from ponds and streams), dredges (Goin 1942), and seines. Small plastic or metal tea strainers are useful for catching larvae in shallow pools. A dip net or two is always useful, a seine is bulky and difficult to transport. A large dip net is often as useful for aquatic herps as a seine. You may want to construct a special filtering device such as the one described by Bragg (1951), a deep wooden frame covered with screen.



Figure 1. Use of funnel trap for snakes, lizards and anurans.

TURTLE TRAPS. A number of different traps for various species of aquatic turtles have been described. For details see Braid 1974; Ernst 1965; Fever 1980; Gordon 1978; Iverson 1979; Legler 1960; MacCulloch and Gordon 1978; Petokos and Alexander 1979; Robinson and Murphy 1975; and Vogt 1980. A discussion of traps and other ways of collecting aquatic turtles (shooting, nets, gaffing, "noodling," set-lines and so forth) may be found in Lagler (1943).

HOOKS, CLAMPS, TONGS AND RAKES. These can all be useful items, but are also difficult and costly to transport. Analyze expected situations carefully before setting out. Clamp-type tongs are very useful for dealing with venomous or aroused snakes (Pillstrom 1954).

POLES. These are used for several purposes. For diurnal lizard collecting, a thin thread noose can be used to pluck lizards off their perches (Medica et al. 1971). By night, many active animals such as frogs can be coaxed onto the end of a pole and then lowered from the upper reaches of the vegetation to the waiting hand of the eager collector. Some telescoping, collapsible types of poles are available, and of course in wooded areas one can always be cut on the spot. Poles are also useful when walking through mud and muck and for knocking animals from high perches. Carpenter (1955) found he could locate turtles in piles of leaves and debris by probing with a thin pole.

NOOSE. As mentioned in reference to poles, many lizards can be noosed if they perch in exposed areas. A loose slip-knot should be made in some very thin line, and attached to the end of the pole. The noose is worked slowly and carefully over the lizard's head, then jerked tight with an upward motion. The lizards may be lured to the noose by using dangling rubber bands (Peaker and Peaker 1967), the noose shot from a noosing gun (Bertram and Cogger 1971), or snap snares set (Stickel 1944). See also Anon. (1971a) and Eakin (1957).

PISTOL. Regulations make carrying firearms rather complicated in most places, but a .22 pistol with a long barrel (6-12 inches), loaded with dust shot (Schmidt 1951) is useful for collecting some very fast moving, diurnal lizards which might be difficult to noose (such as *Cnemidophorus* and *Ameiva*). Some other types of guns have also been recommended (Snyder, 1916).

SLINGSHOT, BEAN SHOOTER, AND RUBBER BAND. These have been suggested as alternatives to a pistol. Slingshots (Engelhardt 1917) take some skill to use and a ready supply of ammunition. Giant rubber bands (Axtell, J. 1965; Brown 1946) are easy to carry, easy to use, and easy to loose. They are about a half-inch wide and six inches long and can pack enough punch to stun most small to medium specimens (and field companions) if fired carefully. Mark your rubber bands with a bright colored pen to help locate them once they are fired gamely at a prey item. Most practitioners of this art prefer to launch the projectile off the thumb, though they can be fired from the outstretched finger or a gun form (Dundee 1950; Neill 1956).

CHEMICAL PISTOL. According to Myers (1956a), small lizards can be dislodged from walls and trees with a water pistol (the "E.R. Dunn method..."). It has been suggested that a formaldehyde solution might be employed instead of just water.

BLOWGUN. Hoddenbach (1966) and Tinkle and Lawrence (1956) described how to make blowguns from metal tubes using corks with protruding needle points for darts. These devices are recommended for collecting lizards during the day.

MACHETE. Very useful in brushy or thickly vegetated situations for cutting trails, poles, and body parts. Machetes can also be used for digging. Take along a file to keep an edge on the blade.

BAIT. Toads and lizards may be attracted to areas where they are easy to catch by using food bait to attract the insects they feed upon (Wilson and Pine 1974). Some collectors have tied insects or fruits by a thin thread to a pole and grabbed the lizards that grabbed the bait (Myers 1956a; Serena 1980).

ELECTRICAL DEVICES. A variety of electrical devices to shock or stun reptiles and amphibians in the water or in the ground have been proposed and variously acclaimed. For details see Anderson and Smith (1950); Gunning and Lewis (1957); Harris (1965); and Joanen and Perry (1971).

SHOVEL. Another item which can take up weight and be hard to pack, but it can be very useful for digging trenches or pitfall traps (see above), and extricating field vehicles from tactical miscalculations. The folding army entrenchment tool is a good compromise, especially if you have an assistant to do the digging.

FIELD KIT. There are a variety of suggested designs for field kits to carry the necessary items needed for preservation, such as the one suggested by Maslin and Swenson (1971). Whatever the shape, be sure it contains the following items:

1. Hardening tray with tight-fitting lid (the first time you spend the night in a stuffy hotel room with a tray full of formaldehyde and a leaky lid, you'll know why...).
2. Paper towels to line hardening trays.
3. Formaldehyde (full-strength).
4. Buffer for formaldehyde (see Fixation, page 13).
5. Field tags, sequentially numbered and with string ready for tying on the specimens.
6. Field book with 100% cotton rag paper, a technical pen with a fine point, and a good supply of appropriate ink.
7. Killing agents (see details below in the section on Preservation of Specimens).
8. Small forceps for positioning specimens in hardening tray.
9. Syringes and needles for injecting killing agents and preserving fluid. Syringe needles may be modified for more rapid injection of fixative by attaching feed tubes or pump-action spray bottles (Congdon et al. 1975; Jackson 1971; Saul 1982).
10. Scissors (small and very sharp).
11. Thread for sewing parts back on specimens, and extra linen thread of the stock used for stringing field tags for use in restringing field tags when you have tied them on the wrong specimens.
12. Liter- or quart-size containers with fluid levels premarked for easy dilution of formaldehyde in the field.
13. Vials with screw-type caps for eggs, larvae, and small specimens. Linda Trueb (pers. comm.) claims that in the days when she was packing into the hinterlands of Panama, she would fill the vials with scotch before the trip, consuming the contents as the vials were needed for specimens. This may account for the abnormally large numbers of tadpoles in her collections.
14. Cheesecloth and plastic bags for packing specimens.
15. Protective handcream or plastic gloves for working with formaldehyde (see Formaldehyde Warning, page 24).
16. Large sewing needles for tag attachment.

CAMERA. Modern 35-mm cameras are now so lightweight, easy to use, and relatively inexpensive that there is little excuse for not properly documenting with photographs any collection worth making. A variety of books offer good discussions and suggestions on how to select and use cameras in ways relevant to herpetological field work. Two of my favorites are by Blaker (1976) and Moldvay (1981). Plan to take along both color and black-and-white film to document specimens and habitats. If possible, two camera bodies (one for color film, one for black-and-white) with interchangeable lenses are preferable. You will also need:

COLOR FILM. It is best to shoot color transparencies. Prints of good quality can be made from color slides, but not good slides from prints. For photographing animals, select a film with an ISO rating near 100. Faster films are a bit too grainy, slower films are hard to work with in many field lighting conditions. Many workers prefer to use Kodachrome ISO 64 due to its durable color dyes.

BLACK-AND-WHITE FILM. Choose an ISO of 125 to 400 for habitat shots, depending on the type of habitat expected. Slower ISO films generally produce better results for this type of use, but light requirements for them can be limiting.

LENSES. In addition to the standard 50 or 55 mm lens for the camera, a wide angle can be useful for some habitat shots, and a short telephoto, macro lens, or 2X teleconverter will accomodate specimen photography. A short (around 35-70 mm) macro-zoom lens will combine all of these features into one lens.

ELECTRONIC FLASH. This makes a big difference in specimen photography, even if it is just used as fill-in for natural light shots. Make sure batteries will be available for it in the area in which you will be working, and take along some spares anyway.

Become familiar with your equipment before departing for the field. Shoot both sample habitat and animal pictures to make sure your light source, lenses, camera and film are all properly coordinated.

When photographing animals, try to pose them on "natural" backgrounds. One trick that will save you a lot of frustration when posing animals is to position the specimen where you want it on the background, and then cover it (Twiest 1968). Compose your shot, remove the covering, and snap the picture quickly. Freshly killed animals may also be carefully posed and photographed. It is also important to photograph the venter of the specimen whenever possible. See Mobbs (1978) and Twiest (1972) for other hints.

TAPE RECORDING EQUIPMENT. The first recording of bird song was made in 1889 (Boswall 1969), but tape recording of frog calls is still ignored by many field herpetologists. Following is a discussion of some types of equipment and field techniques which can be useful.

Recording equipment gets expensive rapidly, so a fair amount of thought as to the eventual disposition of the recorded sounds must go into making your selections. Try to obtain the highest quality equipment you can afford to insure optimum results for your efforts.

RECORDER. Recorders come in two basic formats, reel-to-reel and cassette. Within these categories is a bewildering variety of tape speeds and sizes. A good discussion of what to look for in evaluating the potential field use of a recorder (such

obtuse topics as frequency response, distortion, wow and flutter) can be found in Gulledge (1976). For purposes of making audiospectrographs (also known as sonograms), the main use to which most field recordings are put, cassette format is undesirable. The tape size is quite small and the speed too slow (1 7/8 ips) for really good quality sound reproduction, despite the convenience of the cassette format. Fidelity is related to recording head quality and tape speed. Wider tape means more magnetic particles are used, hence the better quality sound reproduction. The most desirable machine is the reel-to-reel format which takes open reels of quarter-inch tape. The higher recording speeds will produce better results (7.5 ips as opposed to 1 7/8 ips). Full-head recorders utilize the entire width of the quarter-inch tape, which will produce better sonograms than those which use only half the tape (two track). An advantage to machines which record only on half the tape is, of course, that the tape can be turned over and the other half recorded and thus you carry less tape around in the field. Durability of the recorder is another feature to consider. Before purchasing a recorder, try to talk to someone who has used the model in the field to see how it holds up.

Portability of the unit is the next concern. Assuming you have already narrowed your options down to machines designed as "portables," just how easy are they to carry around? From personal experience, I can testify that a Uher Model S, which weighs 8 lbs at the start of the evening, will easily seem like 50 lbs within two or three hours. Be sure to get a wide shoulder strap, and pad it with foam. Rechargeable batteries are a great money saver, but they do you no good when you are away from a reliable source of electricity. Will you be able to purchase the right kind of batteries in the exotic locale you have chosen for recording? In general, look for the following features in a tape recorder for field use:

1. High speed recording capability (preferably 7.5 ips).
2. Recording level meter covering a wide dynamic range.
3. Durability of the unit.
4. Recording monitoring system (speaker and/or headphones) with playback capability (speaker).
5. Good, sturdy case and shoulder strap.
6. Tape counter, to help locate particular portions of recorded tape for playback.
7. Option to take easily purchased batteries.
8. Standard microphone and accessory jacks.
9. Output jack to plug into the sound analyzer back in the lab.

MICROPHONE. It is the microphone which determines what your recorder will have available to record. Once again, a good discussion of this topic can be found in Gulledge (1976). Unlike recording birds, most frogs are relatively easy to get close to without resorting to parabolic reflectors or shotgun microphones, but if funds are available for the purchase of either of these, they can be a big help in the field. See Sinderson (1975) for details on how to make your own parabolic reflector.

TAPE. All tape is not equal. The first consideration is tape thickness, which varies from as thin as 0.5 mil to as thick as 1.5 mil. The thinner tape means more will fit on a reel, which makes it awfully tempting, but it is also easier to break, and tape as thin as 0.5 mil can have sound "print-through", causing unwanted echos of sound. Most people prefer 1.0 mil thickness.

The second consideration with tape is noise bias and equalization. Go for low noise, thicker tape with low distortion ratings. Check these against the tape specifications of your recorder.

Tape Recording Field Techniques

Before you head off to the wilds, don't just know how to operate your recording machine, know how to operate it in the dark. Sinking crotch deep into a leech-infested swamp with the recorder hanging around your neck and the batteries in your headlamp giving out is no time to discover that you can't remember which button is FORWARD and which is REWIND.

Have the following ready when you want to record:

1. Notepad to jot down data which you may accidentally erase off the tape, and to keep your tape numbers straight.
2. Some sequentially numbered tags to drop into the plastic bags with the individual specimens you record.
3. Enough plastic bags to keep each recorded individual separate.
4. Thermometer to determine air and water temperature. This information can be crucial for the interpretation of a tape of a romantically inclined but chilly frog.

Before you begin recording individuals, try to get a good recording of the entire chorus of noises at the site (if any) for a few minutes. This will be useful later to help identify background noises and to encourage the frogs to sing again once you have entered the thick of the chorus to find it suddenly silent (Legler 1964). Whether recording a chorus or an individual, follow these steps:

1. Position yourself in a place where you can stand still and get a clear recording of the sound.
2. Using the recording level meter and monitoring system, check to see that you are picking up the sound(s) you want.
3. Start the tape, holding the microphone as still as possible and shielded from the wind.
4. When the desired length of tape has been recorded, rewind a few inches and play it back to make sure you got what you wanted; then at the end of that segment, record the tape number, date, locality, time, air and water temperatures, species recorded, and identifiable background noises.
5. When recording individual frogs, first collect the individual of merit, then record the information needed on the tape. Bag each recorded individual separately with a tag corresponding to the tape number.
6. Previously recorded tapes of a species can be used in many cases to stimulate a chorus to begin calling, or to start calling again after they have been disturbed (Legler 1964). If this technique is used, it should be noted in the field book with other tape recording information that the call was thus stimulated.

PRESERVATION OF SPECIMENS

Clown (Grave digger): ...A tanner will last you nine year.

Hamlet: Why he more than another?

Clown: Why, sir, his hide is so tanned with his trade that he will keep out water a great while, and your water is a sore decayer of your whoreson dead body.

—William Shakespeare
Hamlet, Act V, Scene 1

Specimens not roughed out for preparation as skeletons or prepared for some special histological technique should be formaldehyde-fixed as soon after capture as practical. Field notes should include a description of the precise methods used for killing and fixing specimens. Include the source of chemicals used and how they were

prepared.

For detailed instructions regarding preservation of reptiles and amphibians, refer to Pisani (1973). A brief outline of the preservation process is presented here.

The amount of care taken with the initial preservation of a specimen is a factor in determining how long the specimen will last. Follow the proper procedures carefully, and make sure that the chemicals used in specimen fixation are fresh and of proper strength. Most collectors carry liquid formaldehyde in a small bottle, but the use of dry paraformaldehyde has been suggested (Huheey 1963; Taub 1962). Sodium hydroxide pellets should be used as a catalyst to synergize the passage of paraformaldehyde into solution instead of sodium carbonate, as the latter seems to promote the rapid clearing of specimens following preservation (Saul 1981).

Killing

Specimens should be dispatched quickly by a method that will leave them relaxed. Reptiles may be killed by injection of Nembutal (aqueous sodium pentobarbital) diluted 1:5 to 1:10, depending on the size of the specimen (Lowe 1956). The specimen may also be snuffed with chloroform, succinylcholine chloride (Lambert 1967), trichloroethylene, a 10% solution of procaine hydrochloride (Livezey 1958), ether (Fowler 1969), or drowned in warm water.

Amphibians are best relaxed in a solution prepared by dissolving Chloretone (hydrous chlorobutanol) (Snyder 1915) in 95% ethyl alcohol, and using a few milliliters of this solution added to a liter of water. Amphibians may also be drowned in warm water. Altig (1980) has suggested application of a small amount of toothache medication containing benzocaine to the heads of small specimens as an alternative means of killing them.

Specimens to be prepared as skeletons by the use of insect larvae should be dried rather than formaldehyde fixed. They may be easily desiccated by hanging them in a wire basket in the engine compartment of your field vehicle (Bond 1939). Amphibian specimens should not be desiccated before preparation (Simmons 1986a).

Fixation

Formaldehyde apparently fixes specimens by denaturing protein by forming cross-links between adjacent protein chains. By thus stopping autolysis and coagulating proteins, the further deterioration of the tissue is prevented (Fink et al. 1979).

Formaldehyde should be buffered because it oxidizes into formic acid which interacts with proteins to form acid solutions (Smith 1947; Taylor 1977). Use of unbuffered formaldehyde will cause problems with later attempts at preparation of material for other types of study because it decalcifies hard tissues.

Taylor (1977) recommends against the use of borax as a buffer for formaldehyde solutions used for long-term storage of specimens, and suggests as alternatives calcium carbonate (in the form of powdered limestone) or a mixture of formaldehyde and glutaraldehyde without the use of a buffer. Quay (1974) has suggested 4 g of monobasic sodium phosphate monohydrate ($\text{NaH}_2\text{PO}_4 \cdot \text{H}_2\text{O}$) and 6.5 g of dibasic sodium phosphate anhydrous (Na_2HPO_4) per liter of 10% formaldehyde. Smith (1947) recommends 200 g of hexamine (hexamethylenetetramine) per liter of full-strength formaldehyde.

Color preservation

Both reptiles and amphibians will fade during storage after fixation in formaldehyde. Several formulas have been proposed which result in better color retention than formaldehyde, but all are somewhat more difficult to use than formaldehyde, more expensive, and require subsequent storage of the specimens in that solution. For details, see Guerra (1976); Waller and Eschmeyer (1965); White and Peters (1969); and Windsor (1971).

Colors of both reptiles and amphibians may also be preserved by removing the skin and drying it. See Beebe (1947); Juszczuk (1952); Kincaid (1948); Paranjape and Mulherkar (1979); Schueler (1981) and Taylor (1981b).

Emergency Substitutions for Formaldehyde Fixation

Sometimes the occasion arises that you have the opportunity to preserve some particularly desirable specimens, but you can't get formaldehyde. In this case, alcohol may be substituted. Slevin (1927) hated formaldehyde, and recommended 65-75% alcohol (ethyl or methyl, although current thinking indicates that methyl should not be used) for amphibians and small reptiles, 70- 80% for large reptiles. It will probably be necessary to make slits in the skin of larger specimens to allow the alcohol to penetrate the tissues, instead of just injecting it as with formaldehyde. This usage should be for emergencies only. Specimens fixed in an alcohol are very fragile, and will often disintegrate if cleared and stained, as we learned from experience at the California Academy of Sciences working with specimens Slevin preserved.

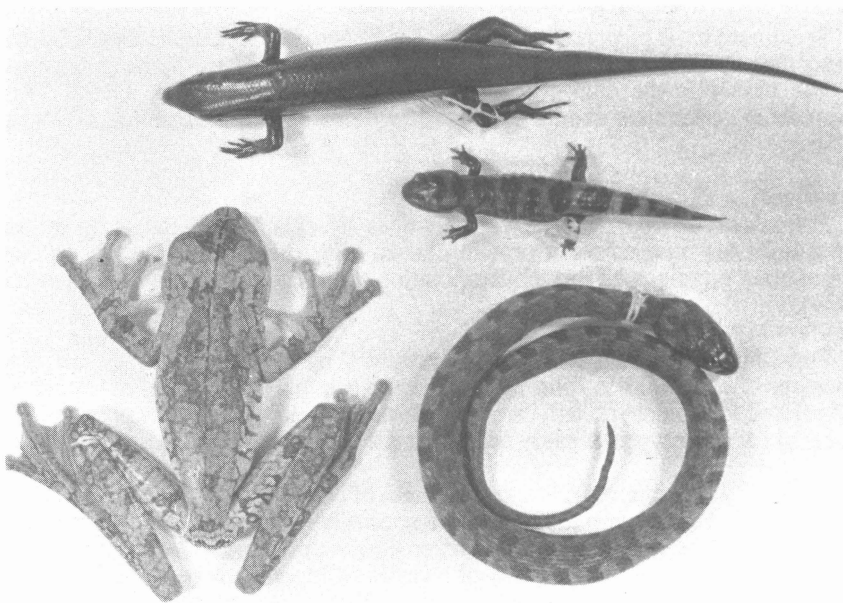


Figure 2. Specimens positioned in fixing tray for hardening.

If isopropyl or ethyl is not available, try a strong, inexpensive liquor. I was once given a collection of snakes from southern Ecuador that a Peace Corps worker had preserved with the local aguardiente (corrupted from sugar cane and flavored with anise). They smelled rather distinctive, but certainly qualified as pickled.

Preparing the Hardening Tray

Select a tray of metal or plastic with a tightly fitting lid and line the bottom with paper towels. Moisten them with a buffered 10% formaldehyde solution.

Preparing the Specimens for Hardening

AMPHIBIANS: Metamorphosed amphibians, except for very large individuals, may be immediately positioned in the preserving tray without injection. Larvae and amphibian eggs are simply put in a vial of buffered 10% formaldehyde solution (Bragg 1949, 1950). When you have amphibian eggs adhering to leaves or twigs, attempt to include some of the piece of vegetation with the eggs. The vial should then be capped and placed on its side so that the specimens are hardened in a flat position.

REPTILES: Reptiles must be injected with a buffered 10% solution of formaldehyde prior to positioning in the hardening tray. For very large specimens, especially when they have been frozen previously and are to be thawed, the use of a 50% formalin solution for injection has been recommended (Johnson and Parker 1980). Inject the preservative in small amounts so that the animal will not become bloated.

1. Lizards. Inject into each limb and the body cavity, and anteriorly into the gular region. Make a series of perforations with a large diameter needle, or slit through the skin of the tail from base to tip on the ventral surface.
2. Snakes. Inject at several points along the body. Large specimens should be injected along either side of the spine, down the full length of the body and tail. One of the pair of hemipenes of male specimens should be everted by injecting the preserving solution into the base of the tail. It is best to sever the adductor muscles prior to eversion. If you do not do this, then when the hemipenis is fully everted, tie a thread tightly around its base so that it remains extended. Make a series of perforations with a large diameter needle, or slit through the skin of the tail from the base to the tip on the ventral surface.
3. Turtles. Extend the head and neck from the shell. Insert a small piece of wood or plastic into the mouth to keep the jaws open. Inject the preservative into the neck, limbs, tail, and deep into the body cavity and lungs (to keep the specimen from floating in the preservative).
4. Eggs. Reptile eggs may be put directly into a vial or bottle of a buffered 10% formaldehyde solution. Large eggs should be injected.

Positioning Specimens for Hardening

Specimens are next positioned in the hardening tray in a way which allows key features of the specimen to be seen readily, the maximum number of measurements to be obtained easily from the specimen, and enables the specimen to be placed in a bottle and packed for shipping without damage to digits or tails (Figure 2). Such techniques as preserving snakes in long tubes (Strayer 1965) complicate future use of the specimens. It has been suggested that animals could be fixed and hardened in plastic bags, ready for shipping (Hahn 1959). This is worth remembering for emergency situations when no preserving tray is at hand, but it is not a good general procedure.

Draw the limbs of frogs to the body, flexed in a natural position, fingers and toes spread apart and straightened to show webbing. Salamanders limbs are flexed and turned palmar surface up on one side of the body, palmar surface down on the other.

Small caecilians are coiled flat to fit into one of the standard size bottles in use in the museum. Larger specimens may be coiled up the inside of a standard size bottle. Lizards are positioned with limbs flexed in a natural manner, toes and fingers straightened, and the tail curved around the body of the animal so that it will fit into a standard size bottle. Small snakes are coiled flat to fit a standard bottle, larger specimens coiled up the inside of the bottle. The neck, limbs, and tail of turtles should be extended from the shell, and the mouth held open by a small piece of plastic or wood. In order to keep the neck properly extended, it is sometimes necessary to suspend the turtle by the head for several hours after injecting it while the initial hardening takes place, or use pins to keep the appendages in place.

Specimens too large to fit in the hardening tray or available bottles may be skinned after measuring so that the body is removed, with the head, tail, and limbs left attached. It may be more desirable to disarticulate the animal and preserve it in pieces (Cook 1965). The skin is then preserved in a tray or bottle, the body prepared as a skeleton if possible. In some cases, it may be desirable to prepare the entire specimen as a skeleton. An emergency hardening tray for very large specimens can be fashioned from a large sheet of plastic, using boards or bricks to hold the sides up, or by lining a pit in the ground with a plastic sheet.

Hardening of Specimens

Specimens positioned in a hardening tray are covered with a layer of paper towels, and a buffered 10% formaldehyde solution is poured in to a depth of several millimeters. The tray is then covered tightly with the lid. Pour the preserving solution over larger specimens coiled in bottles and close them tightly.

Fixing of Specimens

Small amphibians will harden in a few hours, larger ones and most reptiles usually require soaking overnight in the hardening tray. After the specimens have hardened, they should have field tags tied on (sew tags on snakes) and be put in a bottle so that they are completely covered by the buffered 10% formaldehyde. Leave them in the formaldehyde for several days to two weeks at a minimum.

Further procedures and techniques for the preservation and preparation of specimens may be found in Aiyappan and Satyamurti (1960); Altig (1975); Anderson (1948, 1949); Anon. (1899, 1971b); Barco (1976); Bell (1977); Dietrich (1975); Duellman (1962); Elkan (1970); Etheridge (no date); Gloyd (1938); Hower (1967); Johnson (1975); Laughlin and Wilks (1962); Leitao and Alves (1976); Matheus Pozo (1983); McCoy (1929); Mobley (1972); Myers (1956b); Necker (1940); Neill (1950); Nelson (1971); Ortenburger (1923); Pope (1928); Robbins (1976); Tyler (1962) and Walters (1925).

Packing and Transport of Specimens

Whenever possible, specimens should be allowed several days to two weeks in formaldehyde at a minimum before being packed to insure that most of the body tissue has been penetrated. To avoid the problems of transport in liquid and the excess weight of the preserving solution, material may be sent wrapped in cheesecloth dampened with formaldehyde solution, and packed in air-tight plastic bags. Refer to Preparation of Outgoing Loans (page 48) for details of the packing procedure. If the procedure seems like a lot of trouble, imagine what it was like in the "good old days" when specimens were shipped in liquid (Slevin 1927, 1931; Taylor 1950).

When packing, keep the size of the package per plastic bag small. This both makes for easier packing, and reduces the loss to the collection should leakage occur.

Double bag each package, closing the tops of the bags securely. It is preferable to line the interior of the shipping container with a large plastic bag in case of leakage so the container itself will not be damaged.

Ship by the fastest means possible. From foreign countries, this is usually air freight. Surface mail takes from months to forever to reach its destination from overseas, and the specimens will probably dry out. Make sure an address label is included inside the package, and that all necessary customs and postal forms are attached to the outside.

REFERENCES TO SPECIAL PREPARATION TECHNIQUES

Clearing and Staining

Baldauf (1958); Dingerkus and Uhler (1977); Evans (1948); Haber (1964); Hanken and Wassersug (1981); Hardaway and Williams (1975); Lambiris (1971); Mayorya (1965); Russell (1973); Taylor (1967a, 1967b); Taylor and Van Dyke (1985); Wassersug (1976); Webber (1978); and Zug and Crombie (1970).

Skeletal Preparation

Allen and Neill (1950); Applegarth (1977); Banta (1961); Bolin (1935); Borell (1938); Case (1959); Coleman and Zbijewska (1968); Cumbaa (1983); De la Torre (1951); Egerton (1968); Feldman (1980); Gantert (1967); Hall and Russell (1933); Hardy (1945); Hill (1975); Hodlen (1914); Hoffmeister and Lee (1963); Hooper (1950); Hubbell (1958); Hurlin (1918); Jakway and Timothy (1970); Jenne (1974); MacMahon (1961); Maiorana and Van Valen (1985); Mayden and Wiley (1984); Ossian (1970); Patterson and Brattstrom (1971); Rhodin et al. (1976); Russell (1947); Sanders (1953); Simmons (1986a); Sommer and Anderson (1974); Tiemeier (1940); Valcarcel and Johnson (1981); and Vorhies (1948).

Other Techniques

PARASITES: Andrews (1978); Buchanan (1969) CHROMOSOMES: Baker et al. (1971); Becak et al. (1963); Bogart (1968); Branch (1978); Cole and Leavens (1971); Lee (1969); Taylor and Bolanos (1975). TESTES: Bea (1979). BLOOD: Branch (1978); Bush and Smeller (1978); Gorzula et al. (1976); MacLean et al. (1973); Sellers et al. (1980); Sooter (1955); Vosmaas and Zwart (1976). EGGS: Cagle (1944). SECTIONING: Dubois (1942). FREEZING TISSUE: Kretzschmar and Wilkie (1969). STOMACH FLUSHING: Legler (1977); Legler and Sullivan (1979); Taylor et al. (1978). DNA: Sibley and Ahlquist (1981). MISC. BIOCHEMICAL TECHNIQUES: Thorpe and Giddings (1981).

MUSEUM COLLECTIONS

"A major systematic collection of amphibians and reptiles is like a three-way hybrid between a pickle warehouse, a reference library, and a mail-order establishment."

—C.J. McCoy (1981)

A Brief History of Systematic Collections

As the art of magic grew into the practice of science in the western world, odd collections of miscellaneous objects became the ordered systematic resources that today play an integral role in the continuing refinement of our interpretations of

natural phenomena.

The word **museum** originated with the Temple of the Muses founded by Ptolemy Soter at Alexandria in the third century B.C. as a gathering place for learning (Alexander 1979; Bedini 1965; Flower 1898). The plants and animals brought to the Temple of the Muses were the earliest association of formal learning with the collection and classification of specimens.

Picking up interesting things and making a collection of them seems to be a tendency shared by all people (de Borhegyi and Hanson 1982; Ripley 1969). Collections of "curious objects" were assembled for study by such figures as Solomon of Jerusalem, Augustus of Rome, and Aristotle (Flower 1898). Aristotle (384-322) had the advantage of receiving an influx of new specimens from his famous student, Alexander the Great (Flower 1898). The use of students to collect specimens from obscure localities continues even today. There are no records of attempts to preserve and keep specimens in these early collections in the manner that defines a modern museum, but it was from this spirit of "the veneration of the rare, the unusual, the wonderful and the miraculous" (Whitehead 1970, 1971) that museums later arose.

Goode (1889) suggested that apothecaries were the antecedents of the modern curator, referring to the description of the apothecary shop found in Shakespeare:

"...Meager were his looks,
Sharp misery had worn him to the bones;
And in his needy shop a tortoise hung,
An alligator stuffed, and other skins
Of ill-shaped fishes..."

(Romeo and Juliet, Act V, Scene I)

You will note that even then, adequate funding to maintain facilities ("needy shop") was a problem.

With the revival of learning in the Middle Ages, the formal concept of a museum as a place where objects were preserved carefully for study took shape (Flower 1898). The earliest recorded museum devoted chiefly to natural history specimens was that of Conrad Gesner, who was born in 1516 (Whitehead 1970). Upon his death in 1565, his collection was sold to Felix Platten of Basel, where some of it has survived.

In the mid-17th century two developments occurred that were crucial to keeping herpetological specimens. The first was the development of a cheap, clear flint glass (Alexander 1979). Made by using a lead oxide, this glass was more transparent than any previously made. The second development was the preservation of biological specimens in spirits of wine (alcohol). In May, 1622, Dr. William Croone reported on this method of preservation to the Royal Society in London. He probably derived the idea from Robert Boyle (see Boyle 1666), "who two years later exhibited a linnet and a snake preserved in this way" (Whitehead 1970, 1971). This was a far better method of specimen preservation than previous attempts using taxidermy or chemical forms of embalming.

Museums began to flourish towards the end of the 17th century. James Petiver (1658-1718) of Aldersgate in London printed up a sheet of instructions for preserving animals in "Rack, Rum or Brandy" and urged travelers to collect for him (Whitehead 1970). Improvements in methods of preservation seem to have been slow to come. In 1840, Spencer Fullerton Baird, then just 15 years old, wrote to the famous John

James Audubon to describe a bird he shot but was unable to identify. This led to a request from Audubon for specimens, and he instructed young Baird, "Please to collect all the Shrews, Mice (field or wood), rats, bats, Squirrels, etc., and put them in a jar in common Rum, not whiskey, brandy, or alcohol. All of the latter spirits are sure to injure the subjects..." (Herrick 1917).

The 17th and 18th centuries saw an enormous growth in European museums with the influx of strange and wonderful curiosities from the New World (de Borhegyi and Hanson 1982). Many of the specimens acquired by museums in this period are still extant, as testimony to the permanency of properly managed collections.

The Modern Systematic Museum

Linnaeus began to assemble his collection at Uppsala University after his arrival there in 1741. The publication of the tenth edition of *Systema Naturae* in 1758 marked the end of the era of private museums. With the establishment of the system of binomial nomenclature came important public and institutionally affiliated collections as the nature of museums began to change. It has been said that the modern museum "is a product of Renaissance humanism, eighteenth-century enlightenment and nineteenth-century democracy" (Crook 1972).

As the concept of what a museum is has evolved, so has the concept of what constitutes a collection. The nature of collections is undergoing a revolution which places ever more responsibility on a coordinated program of progressive collections maintenance (Irwin et al. 1973). Once the specimen itself and a general geographic provenance were considered sufficient for accession of material. As our abilities to analyze and comprehend the natural world have evolved, our requirements for specimens and data have become more exacting. The importance of individual specimens for their use as voucher material is another fairly recent development (Lee et al. 1982). The minimum standard for localities is far more specific than it once was, and data regarding the relationship of the individual specimen to its habitat are becoming almost as valuable a resource as the specimen itself (Gilman 1923). A modern systematic collection contains not just preserved specimens, but histological preparations, field notes, tape recordings of vocalizations, skeletal preparations, color photographs of specimens in life and of their habitats, maps, and bibliographic resources (Steffan 1977). All these various components of the systematic collection must be cross-referenced for ready access.

Conservation in Herpetological Collections

Conservation of specimens has received little attention in natural history collections compared to art, history, and anthropology museums. There are little data to indicate whether the techniques used in herpetological collections are actually good for the specimens or not. Most techniques and materials are in use today simply because they have been used with apparent success in the past. This is a highly subjective justification at best.

Conservation means preserving each specimen in a way which retains as completely as possible its original composition. Obviously the first consideration for herpetological collections is the nature and quality of the fixative and preservative in use, but there are several important environmental factors to consider, too. Each of these will be discussed in more detail in subsequent sections, but a general overview of conservation problems in herpetological collections is presented here.

The biggest threats to material preserved in alcohol are inadequate fixation, loss of fluid preservative, heat and light (Figure 3).

Unfortunately, the choice of fixatives and preservatives relies more on tradition than empirical knowledge. Most curators believe that formaldehyde fixation is necessary for the long-term preservation of specimens, but formaldehyde has only been in use since the 1890s (Hangay and Dingley 1985). The oldest extant specimens, in various European museums, were preserved in alcohol and have survived nearly 300 years. Very few controlled experiments have been done to determine which is the best preservative, even though formaldehyde, isopropyl alcohol, and ethyl alcohol are all in regular use.

Most workers have observed that specimens do not do well at higher temperatures (Fink et al. 1979). The optimum temperature for storing alcoholic collections seems to be 60-65 degrees F. An increase in temperature means a corresponding increase in the rate of the chemical processes of deterioration (Thompson 1986). At lower temperatures, loss of alcohol from the preserving fluid due to evaporation is also reduced.



Figure 3. Bottles which lost alcohol due to spontaneous cracking of lids. Bottle on the left is now completely dry. Bottle on the right contains mostly water since the alcohol evaporated, allowing growth of bacteria and mold to begin.

Most collections are lit by fluorescent lamps, which give off significant amounts of ultraviolet (UV) radiation. UV is very damaging to specimens, and most workers have observed that specimens kept under fluorescent light for long periods of time bleach very fast and begin to degrade. Sunlight, which has even more UV radiation, is far more damaging to specimens. Lights should be turned on in a collection area only when necessary. The use of some type of UV filters is recommended in collection storage areas with fluorescent lighting. Bottles offer little protection against UV radiation damage, as glass allows UV of 310–400 nm to pass through (Harris 1968; Macleod 1975). No sunlight should ever be allowed on preserved specimens, even through window glass. Incandescent lights produce little UV, but they do produce some heat.

Skeletons, turtle shells, slide mounts, and color transparencies should be kept in closed boxes or cabinets to protect them from dust. Many dust particles are abrasive or acidic (Thomson 1986). Combined with moisture in the air or cigarette smoke (which is very oily), they form a coating on objects that will etch the surfaces it adheres to.

Collections should be protected from fluctuations in temperature and relative humidity. All proteinaceous materials in the collection are affected by these fluctuations—the paper that the field notes, labels, and catalog are written on expands and contracts, breaking along binding seams; skeletal material expands and contracts, teeth fall out of skulls, the emulsion comes loose from photographs, inks fade (Browning 1970; Shahani and Wilson 1987; Thomson 1986). Relative humidity and temperature should be monitored in the collection area over the course the year with a recording hygrothermograph. Once you determine when fluctuations occur and how severe they are, then take steps to ameliorate these changes.

Alcohol preserved specimens are hygroscopic. For this reason, when specimens are removed from their bottles to be examined, they should be kept submerged in a tray of the preservative they are stored in, not in water, as is commonly done. It is easy for the specimens to absorb enough water while being examined that when they are returned to their bottle, they will dilute the alcohol content of the bottle to a dangerously low concentration. Water absorption can also cause tissue damage in preserved material.

Field notes and other permanent collection documentation should be protected in acid-free slip cases or envelopes (Thomson 1986). Binding field notes with acid-free materials is acceptable only if existing holes in the pages are used. Binding methods such as the standard “library binding” where pages are sewn together sideways are not acceptable (Banks 1978). The cellulose of the paper develops a stress line where it is sewn, and with moderate use, will break along this line. When materials in library bindings are opened to be read, the pages are pulled taut against the threads, cutting and tearing the paper. When they are forced down flat on top of a copy machine, the damage is even worse. Even acid-free, 100% rag stock will react to this stress by breaking with handling. Library binding is designed to protect materials from a lot of handling over a short span of time. It is not supposed to protect books well into the future.

Since heat greatly accelerates the deterioration of cellulose (Banks 1978; Browning 1970), the cooler the storage temperature the better for paper, above a lower limit of 60 F (Banks 1978). Recorded tapes must be stored so that the tape is not distorted. Avoid winding the tape too tightly or too loosely by storing the reel after it comes off the machine at playback speed (Wickstrom 1982). Store tape in an area protected

from extremes of temperature or rapid changes in relative humidity. Spools should be rewound every six months to a year to reduce print-through and the uneven tension in the roll of tape that builds up from ordinary environmental fluctuations. See McWilliams (1979) for further discussion of tape and equipment for its storage and protection.

Maintaining Systematic Collections

The daily function of the collections maintenance operation is the continuation of a long tradition, and at the same time an opportunity to initiate innovative improvements. The tradition is the continued care of specimens and data files in the museum. The innovation comes in finding better methods to store specimens, in making access to the specimens and associated data more efficient, and in increasing the quality of the documentation associated with each specimen.

Collections are useless if the specimens sit out the passing decades untouched on dusty shelves. To be valuable resources, they must be made available to the scientific community while preserving their integrity. Data files should be thoroughly cross-referenced and kept up with the addition of specimens to the collection. Physical condition of individual specimens should be monitored continually. The taxonomic status of the species represented in the collection should be kept in accord with the prevailing consensus. Taxonomic changes should be made throughout all cross-catalog references to individual specimens.

In order to maintain the integrity of specimens and data, it is important that standardized procedures be established and documented for the various tasks undertaken in the course of management. Improvements should be incorporated when developed, but standardized procedures insure that different individuals working with a collection will be able to maintain its continuity. Write down the standardized procedures you put into use in your collection, and keep a written record of any changes in these procedures. How you do things today may well be very important to someone using the collection in 50 years. Because you can never predict with certainty when you will be able to resume an interrupted procedure, it is important that it be obvious to another worker what you were doing.

The Future of Systematic Collections

The value of systematic collections will continue to increase in the future. If for no other reason, collection growth will slow owing to worldwide changes as increasing populations expand into previously undeveloped regions. Areas where specimens may be collected without damaging an ecosystem become fewer and fewer as more of the natural areas around the globe are intruded upon and altered by the activities of humans. There also have been changes in the attitudes of people towards scientific collecting. Field work itself has become more complicated and costly in recent years as the expenses of travel and the requirements for permits for specimens compound. All of these factors make existing collections ever more valuable as biological resources and serve as further justification for their protection through careful, thoughtful maintenance.

There was a long period in the growth of museums when amassing large numbers of specimens for the purpose of naming large numbers of species was considered the prime function of systematic collections. However, in recent years, the research based on museum collections has expanded considerably in breadth (Conference of Directors of Systematic Collections 1971; Miller 1985). With proper documentation, preserved collections provide baseline data for ecological studies. Field notes usually contain a wealth of information on the conditions under which the specimens were

obtained. Parameters related to the ecology of a species which are difficult or impossible to measure in the field (e.g., food selection and prey size, reproductive condition) are studied more easily and accurately with preserved material. The role of well-managed systematic collections in the quest for a better understanding of the natural world will certainly continue to expand.

THE COLLECTION FACILITY

Office and Laboratory Requirements

Collection Room

The collection room should not be thought of as a "storage room." Storage rooms are for things which are seldom used, not for housing systematic collections. The collection room should be located so that the curatorial staff has complete control over access to it. It should not have windows. The shelving must be sturdy enough to accommodate the weight of the containers (a one-gallon bottle of liquid can weigh over eight pounds). Moveable shelving (compactors) may be used to save space (Anon. 1976). Another common type of shelving constructed from a steel frame with wooden shelves has been described by Colbert (1961). If shelving is constructed floor-to-ceiling, some sort of ladder or step stool will be needed. Switches should be wired so that lights can be turned on selectively in each aisle so that when one area is being worked in, it is not necessary to turn on all the lights in the room. Ultraviolet (UV) filters should be used on fluorescent tubes (Lull and Merk 1982). Make sure suspended light fixtures do not interfere with access to bottles on the top rows of shelving. Earthquake-prone areas should have restraining bars on the shelves to help keep bottles from tumbling to the floor. Owing to the combined weight of fluid-preserved collections, they are usually housed in the bowels of the museum building; thus, you must take steps to avoid rooms that would be used concurrently for storage of supplies, rooms with inappropriate lighting, or with steam pipes or furnace equipment which would keep temperatures too high. The recommended temperature for a collection room is 68 degrees F or below (Fink et al. 1979). Dust filters should be placed over all air ducts in the collection room, because dust on bottles turns to mud when alcohol is spilled.

Preparation Room

This is the work space where cataloging, loan preparation and receipt, specimen identification, and so forth take place. It should have a sink with hot and cold water, cabinets for storage of supplies, and well-lighted work areas with lab benches or tables and a fume hood adequate to keep formaldehyde and alcohol fumes at a safe level.

Offices

Both the curator and the collections manager need offices which are separate from the preparations area. Often the office of the curator doubles as a research area, and must be equipped accordingly for particular project requirements.

The collections manager's office should have a desk, file cabinets, and typewriter. A small lab bench with binocular microscope is very useful so such chores as specimen identification or special specimen preparation can be carried out away from the common preparations area.

Library

Bibliographic resources adequate for specimen identification are essential. The library for a systematic collection frequently includes materials related to the research programs of the staff. The library also houses such collection documents as field notes, tape recordings, and color slides.

Other Requirements

Catalogs should be housed so that they are protected from potential damage from leaking ceiling pipes, spilled coffee, and other common museum catastrophes. It has been recommended that copies of all written museum records be stored away from the museum so that if a disaster occurs, at least the written data will be preserved (Sutton 1980). If computer cataloging is operative in your institution, arrangements should be made to house a copy of the catalog tapes in a safe place away from the collection. Have the stored copies of the tapes updated from time to time to remain current with additions and corrections to the catalog. Many institutions periodically have traditional catalog pages copied on microfilm for safe storage in another location outside the museum.

Formaldehyde Warning

Anyone who has worked with formaldehyde is aware of the sensitivity to it which usually develops with repeated contact. Many people also react to even small amounts of formaldehyde, displaying such symptoms as irritation of the upper respiratory passages, watery eyes, skin irritation, and coughing. Some people become so sensitive to formaldehyde that they react to the trace amounts found in alcohol containing formaldehyde-fixed specimens. Use of a protective coating of a silicon-based handcream and rubber gloves will provide some protection for the fingers. However, wearing gloves may not offer complete protection from formaldehyde, as it has been shown to penetrate natural rubber, polyethylene, and polyvinyl chloride gloves in a matter of minutes (Raloff 1985). Formaldehyde is also a hazard to soft contact lens wearers. Some of the materials the lenses are made with apparently absorb the formaldehyde vapors, plus formaldehyde vapors can become trapped behind the contact lenses (Cohen et al. 1979).

New evidence has recently been reported concerning the potential carcinogenic effects of formaldehyde (NIOSH/OSHA 1980; Perera and Petito 1982). According to these reports, exposure to formaldehyde vapors has produced nasal cancer in laboratory animals. Work areas where formaldehyde vapors could collect therefore must be ventilated adequately or vapor masks worn (Hearne and Catania 1983). A fume hood is strongly recommended. Museum personnel should attempt to limit their exposure to laboratory formaldehyde. We all are already exposed to it from a wide variety of common products ranging from cosmetics to cigarettes to plastics (NIOSH/OSHA, 1980).

PERSONNEL AND DUTIES

"Such staff as there were [at the British Museum] were overworked and underpaid and often had to take on outside work to support themselves. There were no pensions, so appointees tended to stay at their posts until they died or went mad, which they did with astonishing frequency."

—Lynn Barber (1980)

"These vast collections will spread the elements of Natural Science among the people of New York and the surrounding region, but the quiet workers in the attic, who pursue Science for its own sake, will bring the Museum renown throughout the world."

—O.C. Marsh at the dedication of the
American Museum of Natural History in
1877, quoted by Hellman (1968)

Although the number of personnel will depend upon the size of a particular collection and, regrettably, on the finances of the institution housing it, the following job titles and general areas of responsibility are found in many museum situations.

Curator

The curator has the ultimate responsibility for the collection, its maintenance, and direction of growth (Laub 1985). Usually the curator is not involved in the mundane day-to-day maintenance duties, other than identification of specimens or working up a collection for cataloging (Colbert 1958). The curator oversees research done with specimens from the collection, and attempts to keep funding at an adequate level. The curator should be an individual with a good familiarity with systematic herpetology and should maintain a strong research program.

Curatorial Manager

The collections manager now performs many of the duties formerly assigned to the curator relating to the day-to-day maintenance of the collection, access to specimens and data, and loan activity. The collections manager coordinates the activities of any assistants and is responsible for cataloging and preparation of cross-catalogs, ordering supplies, assisting those who use the collection, and usually cleans out the coffee pot because no one else does. A good collections manager should have an understanding of systematics and be familiar with herpetological literature. Useful skills include typing, technical and grant writing, some mechanical ability, and familiarity with computer use.

Curatorial Assistant

Curatorial assistants are the indispensable drones of the museum world. They labor under the direction of the collections manager, tying tags, topping off the bottles of alcohol in the collection, shelving, filing, and performing assorted other tasks.

ACTIVITIES OF THE COLLECTIONS MANAGER

When engaged in what will be a lengthy project, such as cataloging a large accession, the procedures must be structured to accomodate frequent interruption.

Daily schedule

Certain activities should be scheduled frequently to avoid having a backlog of work hamper the collections management process. These activities include:

1. **SHELVING SPECIMENS IN THE COLLECTION.** Keeping up with shelving will insure an adequate amount of space will be available in the preparation area in the case of the sudden arrival of a large accession or return of a large loan. It will also mean that the maximum percentage of the collection is immediately accessible to a request from a user.

2. **PROCESSING OF CATALOGED SPECIMENS.** Preparation of labels for bottles and cross-cataloging should be kept up as closely with the cataloging process as possible.
3. **TAGS AND FLUID.** A sufficient supply of strung tags, usually several hundred, should be kept available. Keep on hand as well sufficient amounts of mixed alcohol and buffered formaldehyde solutions, bottles, and lids in the preparation area.

Periodic activities

Certain activities must be scheduled at regular intervals throughout the year, including:

1. Ordering the preparation area supplies.
2. Sending out loan inquiry forms for overdue loans.
3. Reporting on the activities of the collections management operation.
4. Checking through the entire collection to make sure that all containers and bottles are filled with the appropriate preservative, that skeletal material is free of vermin, and that the general condition of the collection is good.

ACCESSION OF SPECIMENS

"An overcrowded museum is a healthy museum."

—S.D. Ripley (1969)

Introduction

Accessioning is the process involved from the receipt of material in the museum through its preparation for cataloging (Burns and Rozen 1979; Williams et al. 1977). At the time of accession, the data accompanying a collection should be checked for completeness and clarity to avoid delay during the cataloging process. The collection is assigned an accession number. Usually, all collections received by a museum, regardless of the particular group they represent, are assigned sequential accession numbers by the museum. However, in some institutions each department issues its own series of accession numbers. The accession number is used to identify all correspondence, field notes, permits, and other documentation related to the particular collection, so label such material accordingly.

When deciding whether or not to accession a collection, the following guidelines should be considered:

1. Does the material fall within the categories identified as important to collection growth?
2. Is the material appropriate for the collection, or could it be of more use in another institution?
3. Is the material well preserved?
4. Are field notes adequate and specimen tags in good order?
5. Are the proper permits for collecting, exporting, and importing with the material?
6. Has a clear understanding been worked out with the donor regarding terms of deposit of the collection?
7. If specimens come from a zoo or live-animal laboratory, has proper necropsy protocol (Jacobson 1978) been followed? Do you know if the animals were exposed to toxins, infectious agents, or radiation before coming to you?

DOCUMENTATION OF THE COLLECTION

Field Notes

The field notes prepared by the collector at the time the specimens are taken in the field are one of the most valuable parts of any collection. These notes should be deposited permanently in the same institution as the specimens, and cross-referenced to include catalog numbers assigned to specimens. If it is not possible to obtain the original field notes with the collection, at least get a copy of them.

Permits

The morass of laws and regulations engulfing specimens of most any type of wildlife can be confusing, but make sure all incoming material was obtained legally. Copies of all permits, importation forms, etc. should be kept as part of the permanent record of any accession. Refer to Berger and Phillips (1977) and Berger (1980) for details on some of the legalities of specimen acquisition.

Photographs

Transparencies should be labeled with the specimen's museum number (where appropriate), locality, and name of the photographer. Photographs without specimens to go with them are also useful in the collection. When a large number of slides are accumulated, they should be numbered sequentially and indexed by species for easy access.

Negatives should be kept with a proof sheet and a list of museum numbers (where appropriate), localities, and name of photographer correlated to the frame number on the negative. Prints from negatives should be labeled with the necessary information on the back of the print with a press-on adhesive label.

Tape Recordings

Depending on the number of recordings in the collection, the tapes may be spliced together to fill large reels. A tape number is then used for reference in the catalog.

THE ACCESSION PROCESS

An accession number is assigned to each collection of one or more specimens received in the museum for cataloging into the collection.

After the collection has been approved for acceptance, unpack it with care, watching for loose specimens, ruptured containers, information written on specimen containers, field notes stuck between the packed specimens, or slips of paper containing specimen data tucked in with the specimens but not attached to them. Some collectors can be quite creative when they pack things up. An initial assessment of the contents and condition of the specimens should be made for future reference.

Complete an accession form for the specimens. The form should contain the following information:

1. **SOURCE.** The name and address of collector, donor or seller. Include full names of all collectors involved.
2. **DATE.** The date the collection was received by the museum.
3. **ACCESSION NUMBER.** Sequential accession number assigned by the museum.

4. **DESCRIPTION.** General description of the collection and localities of specimens.
5. **DEPARTMENT.** The department or division of the museum accepting the accession (in some cases, an accession will contain specimens distributed among several different departments, and this should be noted as well).
6. **CONDITION.** Condition of acceptance (gift, donation, purchase, expedition, bribe for admission to graduate school...).
7. **FIELD NOTES.** Whether or not field notes are received with the material, and under whose name the notes are filed. In the case of an accession shared by more than one department in the same museum, record where the original notes are to be housed. Make sure field notes include the full names of all collectors.

SELECTION OF A PRESERVATIVE

Which fluid to use as a preservative has thus far been a matter of tradition, with no qualitative data to validate the selection of one solution over another (Fink et al. 1979). Since the 1600s, when spirits of wine began to be used, the overwhelming favorite of curators appears to have been ethyl alcohol, usually in a 60-75% concentration, followed by the less expensive, easier to obtain isopropyl alcohol and formaldehyde. Methyl alcohol ("Wood alcohol") is unsuitable for use as a preservative.

Just because most fluid-preserved collections use ethyl alcohol does not, of course, mean that it is the best preservative to use. Some formulas have been proposed with a specific function in mind such as preserving the colors of specimens (see page 13) or making specimens useful for specific types of histological techniques (Cleverdon, 1943). Assuming that the main purpose of a preservative is to prevent the bacterial growth which would result in degradation of the specimens, a few other solutions have been tried, among them phenoxetol (pers. comm., Lynn Barkley). There are insufficient data available at this time to evaluate their effectiveness compared to alcohol.

The biggest drawbacks to ethyl alcohol are its expense and the complications of obtaining the necessary permits to purchase it tax-free. According to Fink et al. (1979), many curators feel that ethyl alcohol keeps specimens more "firm" than isopropyl alcohol, and it is more pleasant to work with. Both kinds of alcohol will evaporate quickly from a mixed solution if the jars are not capped tightly, reducing their effectiveness as a bactericide.

It is known that the use of isopropyl alcohol makes the specimens unsuitable for most types of histological preparation (Jones and Owen, 1987). In addition, many people find the odor of isopropyl alcohol objectionable. Fink et al. (1979) further report that since isopropyl alcohol has a specific gravity near that of water, it is difficult to measure the strength of its dilution with a hydrometer. In low concentrations (below 45%) rapid clearing of specimens may result. Most users of isopropyl alcohol use a 40-55% solution. One of the major users of isopropyl alcohol is the ichthyological collection at the Bishop Museum. Following a thorough rinsing and soaking that removes all or almost all of the fixative, their specimens are placed in 55% isopropyl alcohol diluted with distilled water (pers. comm., Arnold Suzumoto).

Transfer of Specimens to Preservative

Collections usually come from the field packed in formaldehyde. Traditionally, specimens have been soaked in water (except specimens which are to remain in

formaldehyde permanently) for 24 hours in open containers. They were then transferred to the appropriate preservative, usually a 70% solution of ethyl alcohol.

Damage can result when fixed specimens are soaked in water. Once the formaldehyde is replaced with water, enzymatic activity in the tissues can reoccur (Taylor 1981a). Because the specimens are going into an alcohol solution with less water than the fixative, and because formaldehyde causes tissues to swell, but alcohol causes them to shrink (Cato 1986), many workers now prefer to stage the specimens during the soaking process from 10% formaldehyde up to the 70% ethanol (or other solution) that the specimens will be stored in (Fink, et al., 1979).

It has been suggested that the specimens not be washed at all, but transferred directly from 10% formaldehyde to the alcohol solution. This presents problems, as the trace amounts of formaldehyde in the specimens seem to cause the acidification of the alcohol solution, which will decalcify the specimens (Dingerkus 1982). Therefore, if direct transfer is used, the alcohol solution must be changed until the pH has stabilized and the formaldehyde is removed (Taylor 1981a). There is a simple spot test that will detect amounts of formaldehyde as low as 1.5% in alcohol solutions (Waller and McAllister 1986).

Unfortunately, at this time there are no empirical data to indicate which of these procedures will do the most to prolong the usefulness of the specimens.

Transfer of Specimens Between Alcohols

Most fluid-preserved collections are maintained in either ethyl or isopropyl alcohol. Do not change specimens on loan from one type of alcohol to another without the permission of the loaning institution.

Changing a specimen from ethyl to isopropyl alcohol causes severe damage to the specimen (Jones and Owen 1987) and significantly alters the body proportions of the specimens (Lai 1963). However, in Lai's study, specimens in the reciprocal test (changed from 40% isopropyl alcohol to 70% ethyl alcohol) showed no significant change in proportional measurements. Lai also gives data which demonstrate that proportional measurements from specimens in one kind of alcohol should not be compared to those from specimens in another kind of alcohol.

Specimens should probably not be transferred from isopropyl alcohol to ethyl alcohol until the effects of this change have been studied in more detail.

Identification of Accessioned Material

After a collection has been accessioned, as part of the process of preparation for cataloging, identifications should be made by the curator or worker designated by the curator. This step greatly speeds the cataloging process and minimizes the possibility of errors which may creep in when secondary catalog entries are necessary to add in names.

CATALOGING OF SPECIMENS

Cataloging is the process by which a specimen is assigned a permanent reference number. This number associates the field data with the specimen, makes it possible to retrieve the specimen from the collection, and is used in all cross-catalog files.

How "fine-tuned" the category files are will depend on the size of the collection and how it is used. Most requests for data from a collection are for information on the geographic origin of specimens or for a list of holdings of a particular species.

A mistake in the cataloging procedure is perpetuated easily in the collection (and often cannot be corrected), so the utmost care must be taken to assure the accuracy of the cataloging process.

Catalogs, Cross-catalogs and Related Files

There are several format options for catalogs, cross-catalogs, and related files:

1. Bound ledger book (pages permanently bound).
2. Loose-leaf notebook or record book.
3. Card file.
4. File folder system (used mainly for loan files, etc.).
5. Electronic data storage (with appropriate print-out kept in binders as needed).

When using hand-written catalogs, there is a distinct advantage to having the sequentially numbered catalog in a bound book—pages cannot be lost from it. The loose-leaf or loose card format is better for cross-catalogs, since frequent additions to these are needed, and they are easier to photocopy.

In terms of access to the data, the least expensive and most efficient system is computerized files (Humphrey and Clausen 1977; Leviton et al. 1982; Manning and Cunningham 1982; and Sarasan 1981), but the initial expense of equipment and system implementation may be prohibitive (especially for the smaller museum). Computers enable you to manipulate records easily for special purposes such as constructing distribution maps (Koepl et al. 1979) or generating lists of locality records to respond to requests for collection data.

For photocopying, the card file is superior to loose-leaf notebook pages, as there is less wasted space (more than one card at a time can be copied on a page). Most collections utilize a mixture of the four basic systems listed above. For instance, the species cross-catalog might be kept in a loose-leaf notebook with standard size pages, the geographic cross-catalog on 5x7 cards, the main catalog in traditional bound ledger books, and some specialized files kept on computer.

The following is a list of some of the kinds of catalogs, cross-catalogs, and related files which may be maintained for the collection (each will be discussed below):

Main Catalog	Cross-catalogs	Files
Sequential (1)	Geographic (2)	Loan activity (1)
	Species (2)	Accessions (1)
	Anatomy (3)	Facilities use (3)
	Type material (3)	Loans to staff (2)
	Tape recordings (3)	Loans by individual (2)
	Photographs (3)	

- (1) Always to be maintained in a systematic collection.
- (2) Useful with increasing size and use of collection.
- (3) Optional depending on collection size and resources.

The Cataloging Process

Once a collection has been accessioned, it should be identified and prepared for cataloging as soon as possible. Until it is cataloged, it should be labeled clearly with

the accession number. Preparations for cataloging include:

1. Determination that all specimens have accurate data.
2. Determination that all localities conform to the standard format (Riemer 1954).
3. Arrangement of the specimens by species within each locality to facilitate catalog entry.

Standard Localities

Localities in the catalog must be located easily on available maps, and should be specific enough to eliminate any ambiguities for subsequent workers. See Riemer (1954), along with R. Axtell (1965) and Hutchinson (1964) for a discussion of localities and standard format. The catalog entry in standard format is written as follows:

COUNTRY: State, Province, District, or Region: Other subdivision if any:
Distance and direction to nearest map locality, elevation

Example:

ECUADOR: Pichincha: Hacienda Espinosa, 9 km W Santo Domingo de los Colorados, 600 m

Abbreviations

In order to enter the maximum amount of information possible in the space provided, abbreviations are commonly used. Be sure to keep a list, card file, or directory of the abbreviations you use. Trying to decipher abbreviations from something cataloged 50 years ago is very difficult.

Map Coordinates

When coordinates are given, they are entered in the following format:

00 00'00"N; 000 00'00"E

Arrangement for Cataloging

The material to be cataloged should be arranged so that within a given locality, all the members of each species can be cataloged sequentially. This method enables the cataloging process to proceed much faster and more efficiently by maximizing the number of data entries which can be continued by the use of ditto marks. In addition, when preparing cross-catalog entries, this will save space and increase efficiency by enabling sequential numbers from the same locality of the same species to be entered as a unit (Williams et al. 1977).

Catalog Entries

Entries in hand-written catalogs should be printed neatly in a stable black ink. See Williams and Hawks (1986) for a comparison of inks. Never use ball point or felt-tip markers. Minor mistakes may be erased thoroughly with a clean, high-quality ink eraser. Larger errors must be crossed out with one line, the correction written in above, and then initialed and dated by the cataloger. Whatever the format of the catalog, the following categories should be included:

DATE OF COLLECTION. Dates are entered as the numerical day, the first three letters of the month and the numerical year, in this format:
day/month/year

Example:

24 Jan 1876

SCIENTIFIC NAME. Scientific names must be approved by the curator before entry into the catalog. If a specific or generic name of questionable validity is provided, the name should be entered in hand-written catalogs in pencil until an acceptable name is determined for that specimen, at which time the pencil

can be erased cleanly and the appropriate name entered in ink. If no scientific name is provided, simply print lightly in pencil the name of the family group to which the specimen belongs. These pencil names may then be erased and an accepted scientific name entered in ink when it is determined.

LOCALITY. Use standard format as discussed above.

COLLECTOR. The initials and last name of the collector or collectors of the specimen. Be sure that the **full name(s)** of the collector(s) is listed on the accession form for the collection.

FIELD NUMBER or ORIGINAL NUMBER. The field number for specimens received directly from the collector or the original museum reference number of specimens received from another institution.

STORAGE MEDIUM OF SPECIMENS. If other than the standard in use in your collection, or if the specimens are stored in some special condition (e.g. dry).

ACCESSION NUMBER. The accession number assigned to the collection the specimen is part of.

REMARKS. This section is used for other relevant information pertaining to the specimen, such as:

1. Type status (Holotype or Paratype).
2. Notation that the specimen number refers to amphibian larvae, amphibian eggs, reptile eggs, etc.
3. Notation that all or part of the specimen has been utilized for a special anatomical preparation, such as a skeleton. Note the sex and snout-vent length of all specimens prepared as skeletons or cleared and stained preparations.
4. Special notes on the time of collection, habitat, etc.
5. Maintenance of specimen alive in the laboratory prior to its disposition in the museum. This is in case someone later wishes to utilize the specimen for some histological study or feeding study that might be affected by laboratory maintenance.
6. Cross-reference to any photographs, tape recordings and so forth of the specimen.
7. Other information as necessary.

Other Cataloging Information

USE OF DITTO MARKS. When the information to be entered in the catalog is exactly the same as that immediately above it, ditto marks are used to avoid unnecessary entries.

LARVAE. Larvae are cataloged together by lot if they were collected together. In the remarks section, enter "larvae."

CROSS-REFERENCE TO FIELD NOTES. The museum number assigned to each specimen should be recorded in the field notes by the cataloger next to the field number. If the museum is not able to retain the original field notes, make sure the museum number is recorded on the museum copy of the notes.

Tagging the Specimens

Once a museum number has been assigned to an individual specimen, attach the sequentially numbered tag to that specimen (see Specimen Tags, page 34):

1. The tag should be tied on the right hind leg, just below the knee, firmly, but not tight enough to damage the specimen.
2. If the right hind leg is missing or damaged, tie the tag on the left hind leg, an arm, or around the waist of the specimen as appropriate.
3. Snakes, caecilians, limbless lizards and salamanders, and amphisbaenids are to have the tag tied around the neck. For large specimens or those lacking a suitable constriction of the neck, the tag should be sewn through the neck

- (below the vertebral column) or through the skin of the neck with a thin needle.
4. To associate numbered tags with animals with limbs too small or too fragile for leg ties, fasten the tag around the waist.
 5. If the specimen is too small or too fragile for a waist tie, place the tag (with about one inch of string attached) and the specimen in a shell vial filled with the proper preserving fluid. Position the tag so that the number can be read through the vial. Close the top with a secure cotton plug (Levi 1966). Avoid leaving air bubbles in the vial beneath the cotton plug. Invert the shell vial in a standard size bottle also filled with the appropriate preserving fluid.
 6. When a number refers to larvae, put the tag (with about one inch of string attached) in a shell vial with the larvae. Make sure the tag is positioned so that it can be read through the vial. Close with a secure cotton plug and invert it in a standard size bottle also filled with the appropriate preserving fluid (usually formaldehyde). Avoid leaving air bubbles in the vial beneath the cotton plug.
 7. Never remove the field tag or previous museum tag from a specimen just because you have added your own museum tag. The previous number is often the only clue available to sort out errors of transcription of data in the catalog (Zweifel 1966).

After Cataloging...

All tagged specimens next are allocated to bottles with proper labels and shelved in the collection room. Cross-catalog the specimen data in the appropriate categories.

CROSS-REFERENCE FILES IN THE COLLECTION

THE GEOGRAPHIC CROSS-CATALOG

The geographic cross-catalog is the arrangement of specimen data by geographic locality. How the geographic cross-catalog is divided (by region, country, state, county, province) will depend on how extensive the holdings are from a particular area. It is kept most efficiently on file cards or in a loose-leaf notebook if electronic data management is not available. File cards or loose leaf pages facilitate rearrangement, photocopying, and expansion of the listings. Use separate cards or sheets for each species within the particular geographic subdivision, and arrange these in the same order as the bottles of specimens in the collection room. If the cross-catalog is computerized, be sure your program can sort by the smallest appropriate geographic subdivision needed for efficient data retrieval (usually county or province).

Entries in the geographic cross-catalog should contain the following information:

1. Family group to which the specimen is assigned.
2. Locality in standard format (as it appears in the catalog).
3. Scientific name.
4. The museum number(s) of the specimen(s) from that locality.
5. If the specimen is a larva, or has been specially prepared (skeleton, cleared and stained, etc.), has a photograph or tape recording on file, or is a holotype or paratype, enter this information.

The geographic cross-catalog should be divided at least by country or some more inclusive regional designation. For countries from which the collection contains large holdings, it is often useful to further subdivide them by state or province.

THE SPECIES CROSS-CATALOG

The species cross-catalog is a listing of the holdings of the collection by taxonomic unit. This cross-catalog is arranged by family group in the same order as in the collection. Within each family group, the listings are arranged alphabetically by genus and species.

The species cross-catalog is best kept as a computer file or in a loose-leaf notebook or on file cards. If the species cross-catalog is to be manipulated via computer, be sure that your program can sort by family group, genus, and species. The specimens in the species cross-catalog should be listed in the order of their museum numbers.

Entries in the species cross-catalog should contain the following information:

1. Family group the specimen is assigned to.
2. Scientific name.
3. Museum number.
4. Locality in standard format, as in the catalog.
5. Date of collection of the specimen.
6. Field number, including initials of the collector, as it appears in the catalog entry for the specimen.
7. If the specimen is a larva, or has been specially prepared (skeleton, cleared and stained, etc.), has a photograph or tape recording on file, or is a holotype or paratype.

THE ANATOMY CROSS-CATALOG

Any specimen in the collection which has been prepared in whole or part as a skeleton, histological preparation, has had stomach contents removed, is cleared and stained, and so forth, should be referenced in the anatomy cross-catalog. The anatomy cross-catalog is arranged most efficiently on file cards or in a loose leaf notebook to facilitate rearrangement and expansion of the file.

Each entry in the anatomy cross-catalog should consist of the following information:

1. Family group to which the specimen is assigned.
2. Scientific name.
3. Museum number.
4. Locality, entered in the standard format as in the catalog.
5. Content (skull, complete skeleton, karyotype preparation, cleared and stained, etc.).
6. Designation of type status, if applicable.

The entries are filed by the family group system in use in the collection. A notation in the Remarks section of the main catalog should be made for each anatomical preparation.

SPECIMEN CITATION INDEX

When a museum specimen is cited in the literature, an entry should be made in the specimen citation index. This file has two main functions. The first is to provide a means for evaluating the use of specimens for exchanges or gifts, availability for loans, or their potential use in some anatomical preparation. The second purpose is for evaluation of the usage of the collection by the scientific community (see Facilities Utilization Records, p. 54). The file should be arranged as is the species cross-catalog. Each entry should contain the following information:

1. Family group the specimen is assigned to.
2. Scientific name.
3. Museum number.
4. Complete citation of the reference to the specimen in the literature.
5. Special remarks as appropriate, such as reference to additional specimens used in a study but not specifically cited by number in the publication.

Specimen Tags

Historically, specimen tags have been almost as varied as the specimens they were attached to (Hawks and Williams 1986). They have been made of paper, wood, metal, plastic and cloth, and carried everything from a simple number to full collecting and locality data for the specimen. The requirements for a specimen tag are as follows:

1. The tag should be relatively indestructible.
2. It should bear at least the museum number, preferably stamped into the tag so that it can still be read even if the ink fades or does not print well, and
3. Be small and light weight so it does not overwhelm or damage the specimen.

Tags should be made of white, 100% cotton stock, with a pH of 6.5 to 7.0 (Hawks and Williams 1986), machine printed in indelible carbon black printing ink (Waters 1936) with the sequential museum number on one side, and the museum name or symbolic code (Leviton et al. 1985) on the other. A small hole should be machine punched into one end for attachment of the linen thread for tying the tag to the specimen.

Tag Vise

To facilitate stringing of tags a clamp or tag vise is useful. This is made from two lengths of wood 12-15 inches long which can be clamped together by two bolts. A series of tags is clamped with the holes outward for stringing. This holds the tags still and frees both hands for faster stringing.

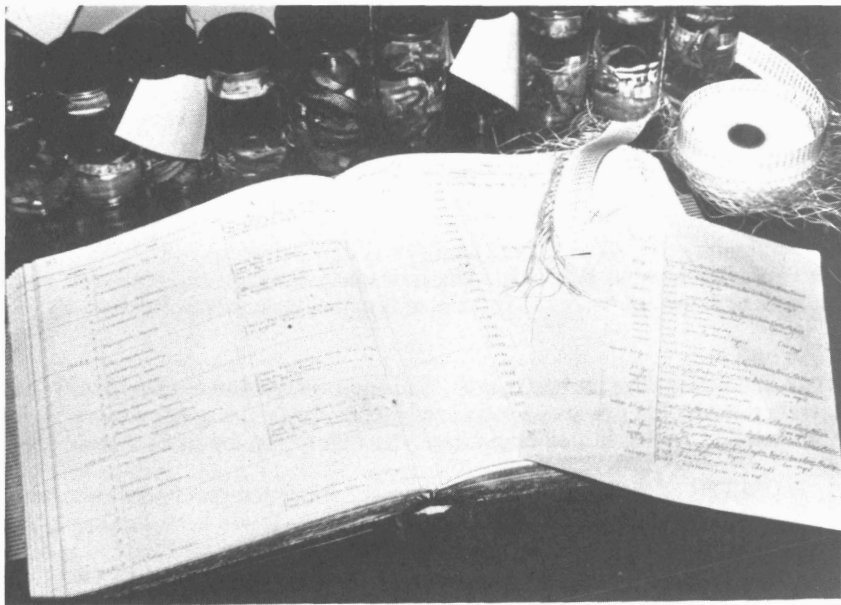


Figure 4. Catalog, specimen tags and field book.

Stringing Museum Tags

Museum tags and field tags are to be strung with linen thread so that they are ready to tie directly on to the specimen. Zweifel (1966) recommends a linen carpet thread, three cord No. 25.

PREPARATION OF LABELS

Each specimen in the collection is kept in a bottle, tank, vial, or box with a label bearing the specimen's museum number, scientific name, and locality of collection. The standard label should be printed on water-resistant, 100% cotton rag paper, preferably nylon-fiber.

Information on the label should be typed neatly with a black carbon ribbon or printed with acceptable ink (see Williams and Hawks 1986). Some of the "correcting" typewriter ribbons have a type of ink which will not bond with some types of papers in alcohol, so be sure to test any previously untried combination of inks and papers before using them in the collection. No correcting fluid or strike-over tape may be used on the labels because they are not alcohol resistant. Mistakes usually can be corrected cleanly with a quality ink eraser.

Combining Specimens in Bottles

To save space, glassware, and alcohol, specimens should be combined reasonably in bottles. All specimens of one species from the same locality can obviously go together unless some specimens are so small that they may be damaged by larger ones in the same bottle. The limiting factors to consider are the number of individuals per bottle and the ratio of specimen volume to preservative volume. Don't lump so many together that it will take forever to find the ones you might want in the jumble. Combining specimens from different localities should follow these basic principles:

1. Never combine specimens from more than one country, no matter how small they are.
2. Do not mix specimens from one geographic subdivision among multiple bottles from a larger geographic subdivision. For example, if you have three bottles of *Bufo speciosus* from Texas, don't scatter specimens from the same county among all three bottles, unless the holdings from one county fill more than one bottle.

Use common sense (if available) so that you don't wind up with a collection all housed in gallon bottles. When deciding how many specimens to put in one bottle, try to stick to a ratio of twice the volume of liquid as specimens (Zweifel, 1966).

Label Content

The standard label for the bottle, tank, vial, or box of specimens should contain the following information (for such specialized labels as those on small vials of cleared and stained specimens, it may be necessary to abbreviate the label content somewhat):

1. **MUSEUM NUMBER.** The museum number of each specimen in the bottle. Non-sequential numbers must be listed individually in numerical order, but sequential numbers for specimens from the same locality are listed inclusively.
2. **FAMILY GROUP.** The name of the family group in which the specimens are arranged in the collection. Some institutions use a system of numbered family groups, putting just the appropriate number on the label. This does make shelving of specimens a little easier for workers unfamiliar with the collection. The obvious advantage of using the family group name instead is that the

frequent rearrangement of the collection with the recognition of new family groups or sinking of old ones is possible without changing all the numbers on all the labels (a horrifying thought to be sure).

3. **SCIENTIFIC NAME.** The generic and specific name of the bottle contents.
4. **LOCALITY.** The locality is entered in the standard format, as it is in the catalog entry for the specimen.
5. **TYPE STATUS.** If the specimen is a holotype or paratype, this is duly designated, usually in all capital letters.
6. **PRESERVING FLUID.** If the specimen is stored in a preserving fluid other than the standard in the collection. Bottles containing formaldehyde should always be so labeled.
7. **TANK SPECIMENS.** If the specimen is too large for a standard size bottle, it is allocated to a tank. In this case, an additional label is prepared but kept dry in the smallest size standard bottle in use in the collection. The bottle is shelved with the other holdings of that species. The label also should have the number of the tank containing the specimen on it.

Soaking Labels

Once the label is completed, it may be necessary to submerge it in a soaking tray of 70% ETOH until the initial ink run has faded. This usually just takes overnight, although the process can be speeded up by frequent agitation of the soaking tray and labels.

Labels for Specimens Stored Dry

Specimens stored without fluid have labels prepared as detailed above, but with the word DRY on them to avoid confusion during alcohol replacement. Labels for dry specimens are not soaked.

Skeletal material stored dry in plastic or cardboard boxes is labeled on both the outside and the inside of the box, with the same information as contained on labels as detailed above, but including also the sex and snout-vent length (SVL) of the specimen. To prepare both labels quickly, type the outside labels on a sheet of press-on type address labels, then photocopy the entire page and cut it apart for the inside labels.

ALLOCATION OF SPECIMENS TO BOTTLES

A variety of standard sizes of bottles should be available for use in the collection. Using bottles of set standard sizes makes the process of replacement of caps and liners easier, as well as making it possible to more accurately estimate shelf usage for expansion and rearrangement of the collection. The recommended standard sizes of bottles for a herpetological collection to stock are half-pint, pint, quart or liter, half-gallon or two-liter, and gallon.

Purchasing bottles in large quantities directly from the manufacturer will greatly reduce the cost per bottle. The nagging problem with bottle supply is that manufacturers have a knack for suddenly ceasing to make your most useful size and shape jar.

A good discussion of the two types of jars currently in use in most collections (screw-cap and bail-top) may be found in Fink et al. (1979), but their chief conclusions are discussed below.

Screw-top Jars

These come in a large variety of shapes and sizes. Straight-sided, flat-bottomed jars of flint glass (clear glass) are the most desirable. Although available from scientific suppliers, they are much cheaper from commercial suppliers, and cheaper yet purchased in quantity from the manufacturer.

Metal lids are not recommended for screw-top jars in use in the collection because of rust. The rusty lids leak, are often difficult to remove from the jars, and the rusting metal chips from the lid can fall on the specimens and discolor them.

The plastic lids traditionally used are made of bakelite, a hard, phenolformaldehyde polymer. There are two main problems with these lids. The first is getting a good seal when the lid is screwed down. Most of these lids come with a cardboard or foil liner which will not withstand collection use. There are three types of liners which seem to be successful in providing a good seal. Two of these are liners which insert into the neck of the bottle and have a flange which provides the seal between the rim of the jar and the lid. The third type is a flat, polyethylene disk, 0.040 to 0.080 inch thick (Palmer 1974). These disks must be cut with close tolerance to the lids in order to provide a good seal. The second difficulty with bakelite lids is that they tend to "back off," or become loose after being tightened down, especially with fluctuations in temperature or with vibration of the jar. Allow them to sit overnight after they have been tightened and then tighten them again before shelving the jars. The bakelite lids can also be screwed down so tightly that they crack and break.

A superior lid is made of polypropylene, a more flexible plastic. The best kind have knurled edges, and with a polypropylene liner can easily be tightened firmly enough to provide a good seal. These lids are quite resistant to backing off.

There is a flexible plastic lid available for gallon jars that also seals well. It has two pieces, a threaded rim with a flange and a large disk which fits inside the rim. If plastic lids for the gallon jars in your collection cannot be obtained, the lifespan and effectiveness of the metal lids can be extended by using a layer of parafilm between the lid and the jar (Peden, 1980), a sheet of mylar, or by coating the inside of the lid with petroleum jelly.

Bail-top Jars

Bail-top jars have glass lids with a rubber gasket and metal clamp. Unfortunately, they are becoming hard to find, and many sizes must be imported from France or Italy.

The rubber gaskets of these jars become cracked and brittle after contact with alcohol and must be replaced periodically. There are a number of other materials that replacement gaskets can be made from that are far more resistant to alcohol than rubber, e.g. a synthetic Nitrile rubber, EPDM (Nordel) Buna-N, butile EPT, and neoprene (Anon. 1980; Fink et al. 1979).

Filling the Vessels

The smallest bottle that will reasonably hold the specimens listed on the label should be selected. The bottle should be large enough that digits and tails of the specimens are not stressed, but not so large that excess fluid and shelf space are used in its disposition. A good rule to follow is to allow approximately twice the volume of liquid as specimens (Zweifel, 1966). Too many specimens in a bottle will dilute the preserving solution significantly (Taylor 1981a).

The label should be flat against the inside of the bottle, without overlapping any part of the specimens.

The bottle should be filled with the liquid preservative to a level between the shoulder and the lid, so that the specimens are well covered. When all the bottles are filled to the same level, those which have lost fluid by evaporation or leakage will be detected readily. In the event that an exceptionally long specimen protrudes beyond the fluid level, cover the protruding portion of the specimen with a layer of cheesecloth which has its edges below the level of the preservative, so that the specimen remains moist.

Bottles containing formaldehyde should say FORMALIN on the label. Types have a colored ribbon tied around the neck of the bottle (usually blue for paratypes and red for holotypes) and PARATYPE or HOLOTYPE on the label.

Some Tricks for Removing Tight Lids

When attempting to open a difficult lid, grasp it so that your hand goes around the edge of the lid as completely as possible. This way, even pressure can be applied all around the edge.

For screw-type lids, try wrapping a wide rubber band around the lid to give a better surface to grip while twisting the lid. The bottle may also be inverted and struck squarely and firmly on a flat surface, or turned slowly while tapping around the edge of the lid.

For bail-type bottles, loosen the clamp at the bottle neck, pull the bail back from the top of the lid, and remove the lid. If the lid is stuck, pull the tab which protrudes from the gasket. When this breaks off, twist the lid, tap the edges of the bottle and lid, or, as a last resort, carefully insert a knife blade between the gasket and the lid to break the seal. Use of forceps for this purpose is common but not officially sanctioned.

Once the Bottle is Ready

Wipe the outside of the bottle and lid clean and dry once the bottle is ready to be placed in the collection. Alcohol left to dry on the outside of the bottle frequently leaves behind a sticky film, or collects dust which then turns to mud.

Vials

A variety of sizes of vials should be kept in stock. Screw-cap vials are used in the field, for storing specimens in glycerine, and for shipping small specimens as discussed elsewhere. Shell vials (straight sided vials without threads for a cap) are used for such things as larvae and eggs.

Tanks

For most herpetological collections, really large tanks are unnecessary, an 18 to 20 gallon capacity being adequate. If your collection has such awkward beasts as large crocodiles or enormous sea turtles, a few larger tanks may be needed.

The best type of tank is of stainless steel construction with a synthetic gasket and clamp-type fasteners for the top. These may be stored on roll-out shelving braced to a wall, or on wooden platforms with casters on the floor. The chief drawback to these tanks is their expense. In an effort to save money, some museums have built fiberglass-lined or epoxy-lined plywood boxes (Dundee 1962), but they do not remain as leak-free as stainless steel tanks. The gaskets used to seal the tanks must

be inspected regularly for signs of aging or deterioration from contact with the preservative. Resetar (1982) has made some recommendations for adhesives to attach the gaskets.

Some institutions use plastic drums or buckets or even ancient ceramic crocks to house tank specimens (Kannemeyer 1973; Williams et al. 1977). These types of containers are extremely difficult to seal properly, and their shape does not allow for efficient use of the collection space. An additional problem with ceramic crocks is that the glaze breaks down over time when they are filled with alcohol, and they begin to leak (Smith 1965).

Legler (1981) has suggested a polyethylene bucket with a tight-sealing lid as a cheap alternative to the costly stainless steel tanks, but Schueler and Aniskowicz (1982) have cautioned that some types of plastic (probably polyvinyl chloride) become brittle after 5-9 years of use and will spontaneously crack; thus, their condition should be carefully monitored.

Should you receive a specimen too large for your existing tanks to hold, you can make an emergency tank with a sheet of plastic and bricks or boards to hold the sides up. Monitor closely for leakage.

General Instructions for Handling Specimens Preserved in Liquid

Specimens preserved in alcohol or formaldehyde, if well cared for, will probably keep indefinitely. However, improper handling, especially desiccation, can easily undo the work of centuries of preservation.

When working with preserved specimens, keep them submerged in a tray of preservative (not water), or covered with a cloth moistened with preservative.

When a tail or limb is broken off of a specimen, it should be tied to the venter of the specimen with linen thread (of the type used for stringing tags). Specimens with limbs broken but still attached need to have the loose part tied to the body with linen thread. Specimens with openings in the body wall large enough for body parts to come free should have the opening bound in linen thread of the type used for stringing tags.

When changing old or contaminated alcohol, rinse the specimens in water (but do not soak them) then place them in fresh alcohol.

When specimens are placed in bottles, they should be positioned in the bottom of the bottle, so that if the fluid level lowers from evaporation or a leak, the specimen will not be damaged until the loss of preservative is severe. Of course, before it gets to the severe point, the bright and alert collections manager will have taken steps to prevent damage to the specimen.

The specimen should not interfere with the placement of the label in the bottle. When returning a large number of small animals to a bottle, put in a few of them, keeping the label properly aligned, and gently shake them down with forceps, then add more, watching for spaces forming between specimens that might prevent some individuals from being nearer the bottom of the bottle than they could be.

GENERAL INSTRUCTIONS FOR HANDLING LARVAL MATERIAL

With few exceptions, larval material is kept in a 10% solution of formaldehyde buffered with powdered limestone (calcium carbonate) rather than the traditional borax (Taylor 1977). The American Museum of Natural History uses a 13:1 solution of buffered formaldehyde (pers. comm., Charles Myers).

See Formaldehyde Warning, page 24.

1. When working with specimens, they should be kept moist in a tray containing the appropriate preservative. Cover with a moist cloth if necessary.
2. Larvae are exceptionally fragile, and their fin structures are easily torn. It is preferable to use a spoon rather than forceps to move larvae .
3. Larvae should be put in shell vials by lot with their museum tag visible. The vial is then filled with the proper preservative, plugged tightly with cotton, and inverted in a standard size bottle filled with the appropriate preservative. Do not use corks to cap vials, as they give off tannic acid in the preservative, which might interfere with future specimen preparation (Levi 1966). Screw caps become loose when submerged in preservative, and thus should not be used either.

PROCEDURES FOR HANDLING SPECIMENS IN GLYCERINE

Glycerine preparations, usually small cleared and stained specimens, are very fragile. Specimens should be allotted to containers individually. The expense of glycerine usually means they end up in screw-cap vials.

In storage, the vials may be stood upright in holes punched in one-inch thick styrofoam blocks (Simmons and Loraine, 1984). The blocks may be cut to fit into available drawers or shelving spaces.

Each vial should have an external label giving scientific name, museum number, locality, snout-vent length, and sex of the specimen.

Add a few crystals of thymol ($C_{10}H_{14}O$) to each vial of glycerine specimens to keep down fungal growth. **WARNING:** Use thymol in a well-ventilated area and avoid breathing thymol dust. Skin contact with thymol causes burns, so do not get it in eyes or on skin.

When glycerine specimens are being returned to the collection, make sure the lid is on tightly, and wipe off the bottle or vial with a damp cloth or paper towel to remove persistent sticky glycerine.

ALLOCATION OF SPECIMENS IN THE COLLECTION

"As far as possible, the collection should be arranged in the same sequence as some generally adopted classification."

—Ernst Mayr (1969)

Specimens in standard sizes of bottles, except types, with proper labels in place, are shelved in the collection room. The specimens should be arranged alphabetically by family group. Within each family group, specimens are arranged alphabetically by genus, and within each genus, alphabetically by species. Specimens identified only to family group level are arranged after the last alphabetical entry in that family group. Specimens identified only to genus are shelved after the last alphabetical unit of species in the genus.

It is helpful to delimit each species on the shelf with a small spacer stick. The stick should be about a half-inch square and the same depth as the shelf. This makes the units of the collection more obvious and thus prevents some common shelving errors. It is preferable not to split a taxon between two shelves unless the material occupies more than one full shelf.

Within each taxon, bottles should be arranged alphabetically by geographic origin of the specimens. This arrangement greatly facilitates the location of material for the preparation of loans.

In areas prone to earthquakes, each group of delimited bottles should be pushed together tightly so that bottles are less likely to topple over.

Following are guidelines for allocation of specimens in other than standard bottles with the regular collection:

HOLOTYPE SPECIMENS. Holotypes should be kept on a secure area of shelving or in an enclosed cabinet. They should be arranged alphabetically by genus and species without regard to family group.

DRY SKELETAL MATERIAL. Skeletons should be kept in small plastic or acid-free cardboard boxes with well-fitting lids, labeled on the outside and the inside. The boxes are arranged on shelves or in trays or drawers in the same order as in the main collection.

TANK MATERIAL. Specimens housed in tanks are allocated tank space as they are cataloged into the collection, thus, unless you have a lot of tanks, many tanks will probably end up with a mixture of family groups and localities. The museum number and scientific name of each specimen going into a tank should be recorded on a master tank list by tank number. A small standard size bottle with a dry label is then shelved in the appropriate location in the regular collection with the number of the tank containing the specimen on the label. As with bottles, do not overfill tanks.

SPECIMENS IN GLYCERINE. Specimens in glycerine should be allocated to a special range of shelving, cabinets, or drawers, arranged as in the regular collection (Figure 5). The cost of glycerine usually means that these specimens are kept in small vials which tend to fall over on the shelf or in the drawer. This problem may be corrected by fitting the vials into holes in a one-inch thick block

of styrofoam to keep them upright. Holes may be punched in the styrofoam using sharpened conduit or an empty shell vial (Simmons and Loraine 1984).

PROCEDURES FOR HANDLING SKELETAL MATERIAL

The preparation and care of skeletal material has received little attention relative to the importance of bones in systematic studies. The attitude still exists among some curators that skeletons should be prepared from specimens with no data, because such specimens are deemed "worthless for anything else." Skeletal material is being utilized now more than ever in studies ranging from systematics to functional morphology. Hopefully, this will result in better consideration and care of skeletal material in herpetological collections.

SKELETAL MATERIAL IN LIQUID. Make a notation in the catalog and cross-catalogs as to the nature of the preparation. Material stored in formaldehyde should be buffered with a base other than borax (Taylor 1977). Small or very fragile specimens are inserted in a shell vial filled with the appropriate preservative, plugged with cotton, and inverted in a small standard size bottle filled with the same fluid.



Figure 5. Vials of cleared and stained specimens in glycerine in styrofoam block.

DRY SKELETAL MATERIAL. Depending on size, this material is housed in trays in skeletal cases or on special shelving. Labels are prepared for the outside and the inside of the plastic or acid-free cardboard boxes containing the specimens. Small bones may be kept in shell vials plugged with cotton and put in with the remainder of the skeleton (shell vials are much lighter in weight than screw-cap vials, and thus less prone to damage the other bones). Very tiny skeletal elements may be enclosed in clear gelatin capsules inside the skeleton boxes. The hyoid or other parts of the animal are often kept wet with the dry skeleton. In this case, make sure that the vials they are in do not leak, and check them frequently for loss of fluid. Vials of liquid are heavy and must be attached to the box with tape so they do not roll around and reduce the remainder of the skeleton to bone meal. It is better to house the vials separately from the dry skeletons to avoid problems of crushing and leakage of liquid.

Plastic boxes have several advantages over the traditional cardboard boxes for the storage of skeletons. The contents can be seen without opening the box, the plastic lids usually fit better than the cardboard lids, the plastic boxes do not absorb moisture, and are acid-free.

MATERIAL STORED IN TANKS

Tanks should be numbered sequentially for ease in the location of specimens. Specimens kept in tanks should be represented by bottles shelved in the appropriate space with the regular collection. The bottle label is kept dry, and in addition to the standard label information, it bears the number of the tank the specimen is kept in.

The contents of each tank should be listed on the master tank list by scientific name and museum number. Specimens should be allocated to tanks with regard to specimen size and weight to avoid damage to smaller, more fragile specimens. An attempt should be made to house similar specimens together in the same tank. The net bags for washing lingerie may be used to keep groups of specimens together in a tank, since these bags will permit good circulation of preservative around the specimens. Do not overload tanks.

HANDLING TYPE MATERIAL

Type specimens are the most valuable material in the collection, and thus should be handled with special care.

The type status of the specimen is noted in the catalog under the REMARKS section. The word HOLOTYPE or PARATYPE is clearly indicated on the label with the specimen, even if the original name for which the type status is applicable has been synonymized. Type status is noted in all cross-catalog files.

Bottles of paratypes are identified with a colored ribbon (usually blue) tied around the neck of the bottle, and for holotypes, a red ribbon is used. If the type is a special preparation (skeleton, cleared and stained specimen, etc.) affix the appropriate colored ribbon to its container.

Paratypes may be shelved with the regular collection, holotypes should be shelved in a special area, preferably in enclosed cabinets.

Specific authorization must be obtained before types may be sent out on loan. Special handling and insurance are usually required.

Each type should be listed in the Types in Collection file, which contains the following information:

1. Scientific name.
2. Literature citation for designation of status.
3. Museum number of the type specimen, locality, etc.

MUSEUM COLLECTIONS MANAGEMENT

"It requires a very unusual mind to undertake the analysis of the obvious."

—A.N. Whitehead, quoted in Curtis and Greenslet (1962)

The basic functions of collection management are:

1. To associate permanently individual specimens with relevant data concerning their collection in the field.
2. To maintain the specimens in optimum condition, preserving the maximum amount of information obtainable from each specimen.
3. To make the specimens and data easily and readily available to qualified workers.

It must be stressed that while technicians are rather ephemeral beings, collections and museums are comparatively eternal. The entries made in the catalog today, or the note scribbled on a piece of paper and stuck in a bottle explaining what happened to the contents must be interpreted after 50 or 100 or more years with the same validity with which they could be read tomorrow. Unfortunately, most mistakes will become part of the collection, too.

The Responsibility of the Collections Manager to the Curator

The curator oversees the direction and growth of the collection, but the collections manager is responsible for the curation, maintenance and conservation of the specimens.

The Responsibility of the Collections Manager to the Collection

The primary responsibility of the collections manager is the proper care and maintenance of the specimens and records. Neglect can result in irreparable damage or specimens hopelessly lost in the shelving area. Even the smallest, most casual errors may be perpetuated for years, and within a short time of their occurrence, are often impossible to correct. The collections manager must keep procedures standardized, and protect the integrity and completeness of data and cross-reference files. The collection manager must keep a record of all chemicals and procedures used in the collection, such things as whether tap or distilled water is used to dilute preservatives, and what steps are followed in moving formaldehyde fixed specimens into alcohol. Such documentation is lacking for most collections, but will be critical when new methods of preservation are developed.

The Role of Collections Management in Service to the Scientific Community

A systematic collection is an irreplaceable resource. Properly documented specimens have intrinsic value to specialists in particular disciplines, but also must

be viewed as interrelated components of the living world. Collections of biological materials represent a moment in the evolution of an ecosystem which can be studied with a depth and thoroughness not possible in the field. It is from the detailed investigations of scores of workers that a general understanding of the workings of the environment arises. Many of the collections in museums today represent not just existing biotas, but also those which are disappearing at a far more rapid rate than they can be studied, such as the flora and fauna of the tropics.

It is presumptuous to think that we may know all of the questions that can be answered by a particular specimen and the data associated with it. For this reason, it is the responsibility of proper collections management to preserve and keep ordered all information associated with each specimen and to maintain each individual specimen in the collection under the best possible conditions. Any individual specimen of a series of specimens may at some time in the future prove to be a key to solving an as yet unencountered dilemma.

In 1971, the Conference of Directors of Systematic Collections pointed to several key elements in the relationship of collections management and the scientific community that are still valid today. These elements are:

1. Collections are training grounds for future biologists.
2. Collections are a data resource.
3. One person can no longer be both scientist and curator. There is a need for professional-level collections managers to handle curatorial matters.
4. Use of computers to handle data management will be a must as collections and collection utilization continues to expand.

RESPONDING TO LOAN REQUESTS

(OR HOW TO AVOID A FEEDING FRENZY BY LOAN SHARKS)

Loan Policy

The curatorial staff should formulate a clear and concise statement of policy regarding loan material, and make sure that each borrower receives a copy of it. A loan policy should cover the following concerns:

1. Specimens should not be removed or transferred from the institution to which they have been loaned without the written consent of the curator from the loaning institution.
2. The borrower assumes full responsibility for the material on loan. If the material is being used by a student, that student's major professor assumes complete responsibility for the student's handling of the material.
3. Copies of the loan form must be signed and returned as requested.
4. Written permission must be obtained before any specimen is permanently altered (dissected, skeletonized, etc.).
5. Specimens must be properly packed to be returned, and the shipment insured, registered, or certified.
6. Any parts removed from a specimen (stomach contents, eggs, etc.) also must be returned.
7. The entire loan should be returned at the same time if at all possible. It should be noted that some institutions prefer having the loan returned in more than one package to protect against loss of the total loan in transit if something happens to a package.
8. All taxonomic changes or reidentifications should be communicated to the loaning institution. Copies of all publications relating to the material should be sent to the loaning institution when they become available.

9. Specimens must be cited in any publications resulting from the study of borrowed material, using the correct standard symbolic code (Leviton et al. 1985) and museum number.
10. Specify the period of time the loan is to be made for (usually six months).

Additional topics that might be covered in the written loan policy could include qualifications for a borrower (institutional affiliation, etc.), use of loaned specimens for display purposes, preparation of casts from loaned specimens, and compensation for any damages to specimens while in the care of the borrower.

The following should be considered when evaluating a request for a loan:

1. **SIZE OF THE REQUEST.** Is the number of specimens reasonable? What will the cost of locating, packing, and shipping the material be? Some specimens (e.g. frogs) are relatively easy to pack and cheap to mail compared to, for example, tortoises. Sometimes requests for excessively large amounts of material are divided into parts, with one installment being sent upon the safe return of the previous part.
2. **CONTENT OF THE LOAN REQUEST.** Has the borrower asked for holotypes or paratypes which can't be sent out of the museum, or specimens of which the collection has only a few representatives?
3. **CONDITION OF THE MATERIAL.** Sometimes the specimens requested are old and fragile, or are of a type which do not travel well (such as skeletal material).
4. **USE OF THE MATERIAL.** What does the borrower plan to do with the specimens? Any dissections, even something as simple as making a small slit to check the sex of specimens, must be agreed to in writing. Is the requested alteration of the specimens acceptable to you?
5. **PROJECT DESIGN.** Is the material really needed by the researcher? If the project was not outlined briefly in the letter of request, this should be done before the request is considered further. This is a judgement that some curators might be reluctant to make, but there is the occasional over-achiever who requests far more material than needed.
6. **TRACK RECORD OF THE BORROWER.** By keeping a borrower record of loan activity (as detailed elsewhere, see page 51), you can evaluate the chances of seeing the loan returned within your lifetime. If a borrower already has a large amount of material out on loan, it is usually a good policy to have it returned before more is sent out. If specimens have been sent back in an unacceptable condition, what guarantee do you have that the researcher will do better next time?
7. **LOCATION OF THE BORROWER.** Especially in cases involving large, hard-to-ship, or very fragile specimens, it is sometimes easier for the potential borrower to come to your collection to examine the specimens than for you to send them out on loan. How far away is the prospective borrower located?
8. **PRIORITY USE OF THE MATERIAL.** Does another researcher already have the material on loan, or has another researcher reserved the first option to borrow it for an ongoing, current project?

Usually when a loan request is denied, the researcher who requested the material is invited to come to your institution to work with it under your watchful eye (unless the request was denied for reasons of Priority Use).

The Loan Form

The loan form should be printed on standard size paper (8.5 x 11 inches) and consist of three duplicate sheets. Carbon paper may be used to produce the two copies

needed of the original. The self-carboned forms used by many museums have a type of ink which runs and fades badly when wet, and the copies of the form are frequently splattered with fluid while unpacking a loan. Worse yet, the copies fade rapidly with age (Kortlucke 1981). When loan records are in an electronic data file, it is often more efficient to generate loan forms by computer.

The loan form should contain the following information:

1. Name and address of loaning institution.
2. Name and address of borrower.
3. Date loan is sent out from museum.
4. Means by which loan is to be shipped.
5. Value of loan.
6. Name of technician who packs loan.
7. Initiation of loan.
8. Loan number.
9. Museum number and scientific name of each specimen.
10. Date loan is to be returned.
11. Signature of recipient and date loan is received.

Categories of Loans

There are four categories of receipt and preparation of loans:

1. Preparation of loans of museum material for an outside borrower.
2. Receipt of loans of museum material returned by an outside borrower.
3. Receipt of loans from another institution or individual made for the use of a member of the museum staff.
4. Return to sender of loans made to a member of the museum staff.

PROCEDURE FOR THE PREPARATION OF OUTGOING LOANS

Selection of Material

Material requested for an outgoing loan must be approved prior to packing the loan for shipment. Make sure the curator is aware of any paratypes, holotypes, or unique specimens included in the request. Once approved, the material is retrieved from the collection and brought to the preparation area.

Completing the Loan Form

Information included on the loan form consists of:

1. Name and complete mailing address of the recipient. Student requests must be approved by that student's major professor, and the loan form made out in the name of the professor.
2. Date loan is to be sent out from the museum.
3. Means by which the loan is shipped (Parcel Post, Air/Other, Air Freight, UPS, hand-carried, etc.). This is so the shipment may be traced if it does not arrive safely at its intended destination.
4. Value of loan (for insurance purposes only). For international shipments, loans should be declared to have NO COMMERCIAL VALUE, to avoid the recipient being stuck with enormous customs duty.
5. The name of the technician who packs the loan (woe to those using plastic bags that leak).
6. Initiation of loan (whether at the request of the receiver, request of the sender, etc.).

7. A sequential loan number. Some institutions prefer to use the date the loan is initiated and the name of the individual the loan is made to instead.
8. Museum number and scientific name of each specimen, along with the locality and date of collection, as recorded in the catalog. The museum tag of each specimen should be checked carefully to make sure that the proper specimens are being shipped. NEVER trust the contents of the bottle to match what is on the label. Enter the total number of specimens in the loan below the listing of the museum numbers.
9. Note what preservative is used, and state that the specimens must be maintained in that preservative.
10. Date the loan is to be returned to the museum. Usually, this is six months from the date of the initiation of the loan.

Separate the sheets of the loan invoice. Handle them as follows:

ORIGINAL and SECOND COPY are sent to the borrower in an envelope separate from the specimens. The borrower signs the ORIGINAL upon receipt of the loan, and sends it back to the museum, retaining the SECOND COPY with the specimens.

The THIRD COPY is kept in the active loan file until the material is safely and eventually returned.

When the SIGNED ORIGINAL is returned to the museum, clip it to the THIRD COPY of the invoice in the active loan file.

Upon return of the loan by the borrower, the THIRD COPY is signed and sent to the borrower as the receipt of the completed transaction. The ORIGINAL of the loan invoice, containing the signature of the borrower, is put in the permanent file of completed loans.

Packing Material in Liquid Preservative

When dealing with alcohol and formaldehyde, wrap each specimen in at least one layer of cheesecloth moistened with the preserving fluid. Several specimens may be rolled up in one length of cheesecloth. Keeping in mind the size of the packing containers available, place the specimens together on a length of cheesecloth so that toes, fingers, and tails will not be stressed (Figure 6). Fold over the edges of the cloth and roll the package up securely but not tightly.

The wrapped specimens should be dampened with the preserving fluid and placed in a plastic bag closed tightly to prevent leakage. The first bag is then sealed and inverted in a second plastic bag. A label identifying the contents and requesting return to sender may be placed face-out between the bags.

Plastic bags should be heat-sealed or be long enough to knot tightly. Do not use rubber bands or string to close bags. Rubber bands become brittle quite quickly in alcohol, and string tends to stretch when wet.

Many commercially available plastic bags do not have a seal of sufficient strength for shipment of specimens. The best solution is to purchase plastic bags in bulk as a long roll of 6-12 inch wide, 2 to 3 mil (0.002-0.003 mm) plastic tube, as first suggested by Loveridge (1952). A heat sealer with a one-eighth to one-quarter inch wide band can be used to make good, liquid-tight bags.

In any case, do not leave standing liquid in the plastic bag of cheesecloth shrouded specimens. Rather, thoroughly moisten the cheesecloth and drain the excess. Before sealing, remove as much air as possible from the bag as this will reduce the chance of a puncture or rupture.

The plastic bags are then packed in a suitable shipping container (a can or sturdy cardboard box), supported by some packing material such as plastic peanuts to prevent the bags of specimens from shifting around during transit.

Enclose a return address label inside the package. Label the outside of the container, then wrap it in plain brown wrapping paper, and secure with nylon strapping tape (string tends to get caught in the voracious bulk sorting machines, resulting in the ingestion of the package). Attach the outer address label well.

Packing Material in Glycerine

Specimens in glycerine are generally quite fragile. They should be fitted into the smallest screw-cap vial or bottle into which they can reasonably be packed. The vial or bottle should be completely filled with glycerine, and tightly capped. Wrap the cap to the glass with masking tape or strapping tape to prevent it from working loose during transit. Seal the vial or bottle in a plastic bag in case of leakage, pack in a shipping container padded with suitable packing material, and label properly. Hoffmeister (1973) has suggested packing vials in holes cut in blocks of styrofoam for shipment.

Packing Skeletal Material

Pack bones and shells in a small box with layers of soft tissue paper. Secure the lid of the box with tape, and attach the return address label to the box. Pack the box into an appropriate shipping container, wrap, and label for shipment.

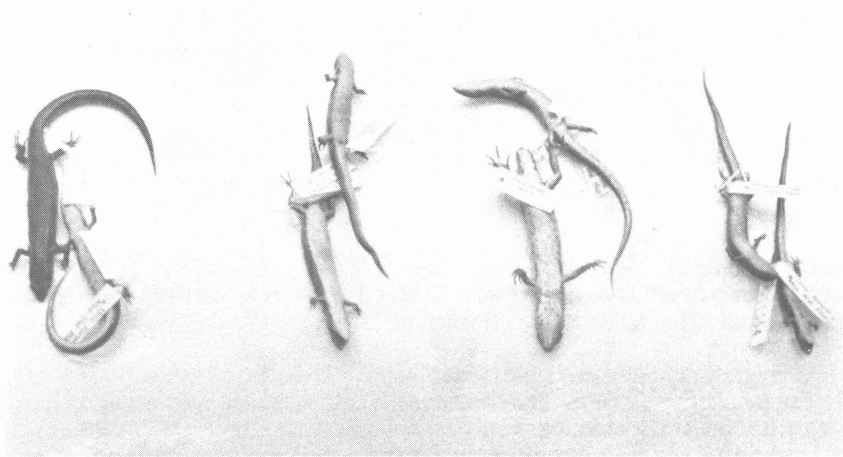


Figure 6. Specimens should be laid out on cheesecloth so that when they are wrapped up, digits and tails will not be bent and tags will not damage specimens.

Shipping the Loan

Domestic loans are sent parcel post, insured (usually for U.S. \$200), or sent by private carrier. Avoid shipping loans during the Christmas rush, from about 15 December to 10 January, as the high volume of packages at this time will at a minimum delay the arrival of your package, and may needlessly endanger it.

International shipments are sent uninsured unless specific instructions for insurance are received. This is to prevent problems with payment of customs duty in the country to which the loan is sent. International shipments under two pounds may be sent "Air/Other." Those over two pounds are usually sent International Parcel Post. Foreign shipments must have the appropriate customs forms attached, with the value of the package listed as "No Commercial Value." The contents of the shipment should be listed as "Preserved natural history specimens for scientific study only."

Marking Bottles of Specimens on Loan

Bottles with the labels in them should never be loaned out with the specimens. The bottle and its label are crucial as a "place holder" and a record of the specimens on loan.

Bottles that have had some or all of their contents removed for a loan must have a loan slip attached before being returned to the shelves. The loan slip should contain the following information:

1. The museum number of each specimen removed.
2. Loan number (or name of individual and date loan is initiated if this system is in use).
3. Initials of person who actually prepared the loan.

Attach the slip to the bottle, and return the bottle to its place in the collection area.

Similarly, vials for cleared and stained specimens, skeletal boxes, and tanks should be marked with a loan slip when specimens are removed.

Records of Loan Borrowers

A file should be kept of loan activity by borrower. Each listing should contain the name and institutional affiliation of the borrower, the dates each loan was sent out and returned, and the total number of specimens involved in each transaction. This information is used to evaluate requests for material on an individual basis.

Nomenclatural Updating

Re-identification of specimens, synonymy of previously applied names, and the naming of new taxa are the three principle reasons for nomenclatural changes. The following procedure should be followed:

1. Obtain authorization from the curator to make the change.
2. Check the contents of the bottles against the labels to make sure that all needed specimens have been located.
3. For hand-written catalogs, make the changes in pencil. Cross out the previous name with one pencil line, and enter the updated name above it. For computerized catalog files, it is usually necessary to delete the previous name from the file to enter a new one.
4. Change the name in all cross-catalogs and other files where appropriate.
5. Prepare new bottle labels with the updated name, and prepare the bottles for return to the collection.

6. Paratypes that have been synonymized are identified on the labels as in the following example:

GASTROTHECA RIOBAMBAE

(Paratype of

Gastrotheca cavia)

7. A holotype specimen retains the original name even if it is no longer valid.

Mixing a Working Supply of Formaldehyde

A 10% mixture of buffered formaldehyde and water is used for fixing fresh specimens and for storage of most eggs and larvae of amphibians.

Mix about a quart of working solution at a time unless a large supply is needed immediately. Use a ratio of 1 part full strength formaldehyde to 9 parts water. If purified water is not available, tap water may be used, but usually a cloudy precipitate will form, so the work solution must be decanted carefully. See the discussion of buffers and neutralizers for formaldehyde solutions on page 13.

Mixing a Working Supply of Alcohol

Acquisition and use of ethyl alcohol is controlled rigidly by the Federal government. The museum must adhere to strict rules regulating the use and dispensing of alcohol to avoid having to pay large fines and/or forfeiting its license to handle ethyl alcohol.

Mix the alcohol with water to make a 70% solution. Allow the mixture to settle for 24 hours before use. If purified water is not available, tap water may be used, but usually a sediment forms in the bottom of the container of mixed alcohol. The use of distilled or deionized water to dilute alcohol is also thought to reduce the initial acidification of the alcohol solution (Dingerkus 1982).

For collections using isopropyl alcohol, a variety of concentrations have been suggested, ranging from 30% to 55%. The Bishop Museum has used a 40% solution of isopropanol since 1965, and a 55% solution from before 1975 for its fish collection with good results (pers. comm., Arnold Suzumoto).

The last of the alcohol mixture remaining in the container of working solution must be decanted carefully and filtered before use (filtering through several layers of cheesecloth is usually sufficient). A hydrometer or density meter should be kept on hand for testing alcohol solutions of questionable spirit.

Alcohol Replacement

Periodically, each individual bottle in the collection must be checked to locate those from which preserving fluid may have evaporated. For large collections, this task is best carried out over an extended period of time in short work sessions (e.g., certain shelving sections can be checked each month). One reason for filling each bottle with fluid up to the shoulder regardless of how many specimens it contains is to detect those bottles which have leaking closures before damage occurs to the specimens. While checking fluid levels in the collection, also look for cracked or broken bottles, lids, and gaskets, improperly allocated bottles, and check the general condition of shelving in the collection room. Bent, cracked, or otherwise damaged gaskets or lids should be replaced immediately when encountered.

When the level of fluid has dropped in a bottle, you must check the strength of the remaining alcohol, since alcohol evaporates much faster than water. Simply adding more mixed preservative will result in specimens being kept in weak solutions, which will affect their long-term survival.

When going through the collection to check for leakage, take along a cart containing a hydrometer or density meter, replacement alcohol and formaldehyde, extra lids and gaskets, and towels to wipe off the bottles before they are returned to the shelves. Long forceps are also useful to reposition specimens and labels improperly placed in bottles.

A procedure to bring some economic efficiency to the process has been suggested (Schleifer 1982). "Old" alcohol is drained through a sieve or filtered through cheesecloth into a 5-gallon container. Full-strength alcohol is then added to this and the solution checked for the proper dilution with a hydrometer or density meter. Thus, the old alcohol is recycled, resulting in a uniform alcohol solution to use in topping off bottles and tanks.

Check tanks for the condition of the gasket as well as loss of alcohol, as most gasket material used in tanks deteriorates from contact with the alcohol. See Resetar (1982) for a discussion of gasket adhesives.

Dehydrated Specimens

Whether or not to attempt the rehydration of fluid-preserved specimens that have become desiccated is highly controversial. Some techniques seem to work, but no studies have been published indicating what effects the processes of dehydration and rehydration have on the specimens.

The most common method has been to soak the specimen in a solution of 30% ammonia and water for a few hours to a few days, then transfer it to alcohol. Another method was suggested by Cleave and Ross (1947). They found that a 0.25 - 0.50 % solution of commercial grade trisodium phosphate worked well with damaged invertebrates and on a large number of vertebrates. Their soaking times varied from an hour for small specimens to two days for larger ones.

In some cases, the specimen may be so dehydrated that it is best just to leave it dry, or a specimen may be so unique that it is not worth risking the attempt to reclaim it. In these instances, be sure the word DRY is on the label with the specimen so alcohol will not be added, which would dehydrate them still further.

Records of Collections Management Activities

Record keeping is essential for the evaluation of the effectiveness of the collections management program, as well as for documentation and evaluation of utilization of the collection by the scientific community. Maintain up-to-date files and cross-catalogs.

Annual Reports

At the end of each year, prepare a general summary of the activities of the collections management program. Include the following information:

1. Summation of the major activities of the year, including ongoing regular activities (such as cataloging and alcohol replacement schedules).
2. Activities of assistants.
3. Loan activity (loans initiated and loans returned, and loans received or returned for staff members). Include the date, name of borrower, institutional affiliation, geographic area of specimens in loan, families and genera of specimens in loan, and total number of specimens in the loan.
4. Exchanges and gifts.
5. Accessions. Include accession number, geographic area of specimens in the

accession, a general description of the content of the accession, and the total number of specimens involved.

6. Cataloging (total number of specimens cataloged, with a list of number of specimens by geographic area).
7. Outside activities of collections management personnel relating to the museum (meetings attended, papers published, public education activities, etc.).

Facilities Utilization Records

Contacts in person, by telephone, or correspondence which require the curator or collections manager to respond in a professional capacity should be recorded as part of the facilities utilization record. This information is needed to evaluate collections resource use. The form should record the following information:

1. Name of person making the contact
2. Address
3. Date of contact
4. Nature of contact and facilities used (library, collections, technical advice, etc.)
5. Member of curatorial staff responding to contact

All visitors to the collection should be asked to sign a visitor book.

Specimens Lost, Exchanged, or Given to Other Institutions

When specimens are known to be lost (usually in the mail), destroyed, exchanged, or given to another institution, this should be noted in the catalog and appropriate cross-catalogs. However, the records for these specimens should still be carried as if the specimens were present. Record the new museum number with its symbolic code (Leviton et al. 1985) in the catalog and cross-catalogs of specimens exchanged or given away. Do not re-use catalog numbers of lost, destroyed, or donated specimens.

GUIDELINES FOR COLLECTION GROWTH

"Peale even preserved an unusual fish without a head, deeming it worthwhile to preserve that much until more could be learned."

—C.C. Sellers (1980)

The philosophy of collection growth was once quite simple—more was better. Any specimen from anywhere was fair game for accession. If for no other reason, the rising costs of housing and data management for large collections have made curators turn away from that attitude, but there are also ethical and legal considerations involved in collection growth. See Cato (1986) and Fritts (1976) for a discussion of museum acquisition policies.

The following criteria should be carefully evaluated on a case-by-case basis when presented with the opportunity to accession new material:

1. Documentation of the Collection.
Does the collection come with adequate supporting documentation in the form of field notes, photographs, and tape recordings? Is the collector reliable? Were the specimens properly preserved and labeled in the field?
2. Do We Need It?
Will this accession complement material already in the museum or fill gaps in the collection? If the accession would be of little or no use to workers in the

museum, are there other institutions which might benefit more from its accession? Is it possible to hold the collection uncataloged for use as exchange material? If the museum already has adequate holdings from the area the accession represents, is there type or voucher material in it requiring deposition in an established institution (Lee et al. 1982) or other circumstances that outweigh the consideration of redundancy? Can the museum afford the costs of acquisition and maintenance of the collection?

3. Is the Collection Legal?

A myriad (some say maze) of regulations confront the collector today both in the country (or state) of origin of the specimens and in the process of getting foreign specimens into the United States. The burden of proof of the legality of a collection rests with the institution receiving it (see Berger and Phillips 1977, and Berger 1980 for a full discussion of the Federal regulations concerning such matters). Is the collection adequately documented with copies of permits and related documents to prove its legality?

4. Ethical Considerations

Once the criteria above have been considered, the curator must ask if the accession of this material is ethical. Does it represent the last chance to preserve the remnants of a vanishing ecosystem, or does it lend encouragement to the destruction of wildlife from a threatened area? Is the collection of more specimens of these species justified just because you don't have them in your museum, or might there already be adequate representatives available in other institutions? Many times the ethical considerations are so subjective as to find no general agreement among members of the scientific community, but they will have an increasingly profound effect on the direction of growth of scientific collections.

Guidelines for Exchanges

There are two types of exchange agreements. The one-time exchange is worked out with specific numbers of individuals of certain species in mind. The open exchange is a supposedly ongoing process that is much harder to maintain. Often museums will establish an open exchange of paratype material in order to have the limited type material deposited in more than one institution in case of disaster. The following considerations must be taken into account when entering into an exchange agreement:

1. No exchange is complete until both parties have the material in question in hand to examine and approve.
2. The paperwork on the exchange should spell out the conditions and terms of exchange exactly.
3. Exchange papers should have space for both parties to sign upon transfer of specimens.
4. Copies of field notes and other data relating to each individual specimen should be included with the specimen in the exchange.

Collections Management Program

The design of a collections management program must be carefully tailored to the particular collection in mind. As a guide to the development of a collections management program for your institution, the following list of criteria is provided. Discussion of the details of specific subjects may be found in the appropriate place in this text. For further discussion of collections management policy, refer to Malero (1979) and Wake et al. (1975).

A. Basic Requirements of a Collections Management Program

1. Accession policy
2. Established cataloging procedure

3. Loan policy
4. Established progressive maintenance schedule
5. Record management system
- B. Other Useful Collections Management Options
 1. Information retrieval system to sort by:
 - a. Geographic origin of specimen
 - b. Taxonomic units
 - c. Anatomical preparations
 - d. Other supporting material (tapes, slides, etc.)
 2. Collection Use Evaluation Files
 - a. Facilities utilization
 - b. Loan activity
 - c. Types in collection
 - d. Specimen citation index

How to Evaluate the Collections Management Operation

Periodically, at least every year or so, it is wise to pause and evaluate the effectiveness of what you are doing, in order to strengthen the collections management program (Simmons 1986b). The following aspects of the operation should be considered:

1. Does cataloging maintain pace with the incoming accessions?
2. Are specimens allocated to bottles and shelved properly in the collection on a regular basis?
3. Is the preparation of cross-catalogs keeping up with the rate of cataloging?
4. What is the turn-around time for loan requests? It should be no longer than two weeks from receipt of a loan request to a response to it.
5. How long is the response time to requests for data? If some types of requests seem to take a long time to fill, or if you are unable to fill some types of reasonable requests, do you need to establish additional cross-catalogs or redesign your information retrieval system?
6. Has the growth of the collection met expectations?
7. Do the curatorial personnel stay busy? Do they have too much to do so that they are continually lagging farther behind?
8. Does the scientific community know that you exist? Evaluate user access records to see if some features of your collection are being overlooked. If so, what steps can you take to correct this oversight? Were you able to respond to the needs of visiting researchers over the past year?

The nature of a collections management program is progressive and dynamic. Frequent evaluation will help assure that your program is able to respond to the changing demands for access by the scientific community, and continue to improve in efficiency.

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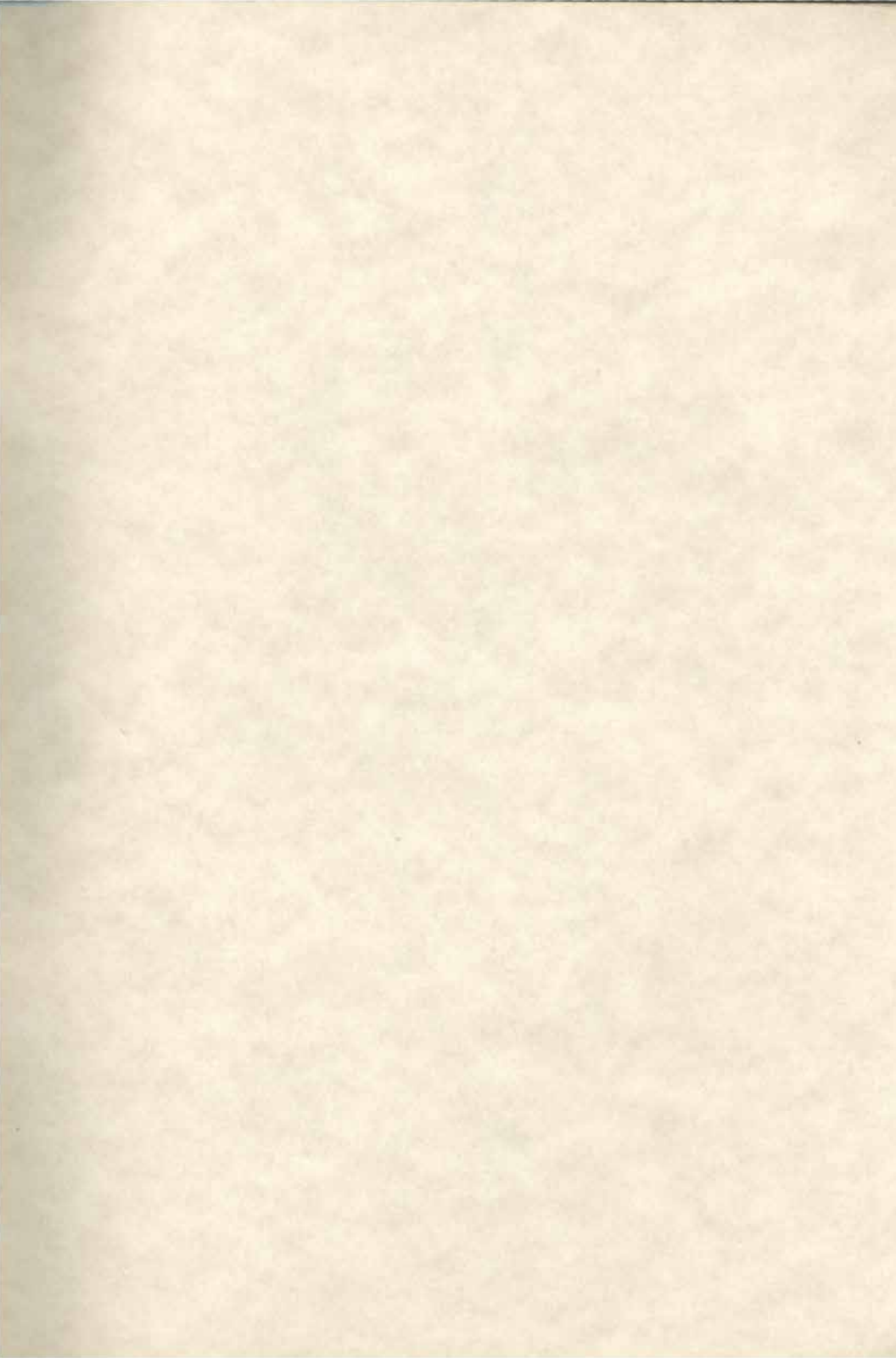
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